Guidelines for Collecting High Priority Ephemeral Data for Oil Spills in the Arctic in Support of Natural Resource Damage Assessments

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Final Report
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Introduction

The Alaskan Arctic region, including the Bering, Chukchi, and Beaufort Seas, faces changing conditions that increase the likelihood of a marine oil spill. Natural Resource Damage Assessment (NRDA) is a legal process conducted by State and Federal natural resource trustees in the event of an oil spill that impacts public resources. Under the NRDA process, the trustees identify and quantify injuries to resources and determine the types and amounts of restoration required to restore those resources and compensate the public for interim losses.

The first phase of the NRDA process evaluates the extent and severity of resource injuries. This phase generally includes collecting time-sensitive, ephemeral data to assess exposure to oil chemicals and effects of those exposures. Ephemeral data are collected in the initial days and weeks of a spill to document conditions that change rapidly over time following an oil spill. Collecting ephemeral data is especially challenging in the Arctic and requires knowledge and methods that are appropriate for the environmental conditions and unique habitats, organisms, and human use resources in the region. High-priority ephemeral data needs for oil exposure and injury assessment were identified through the development of a conceptual model for oil exposure pathways and effects, and consultation with NRDA practitioners and Arctic experts (Figure 1).

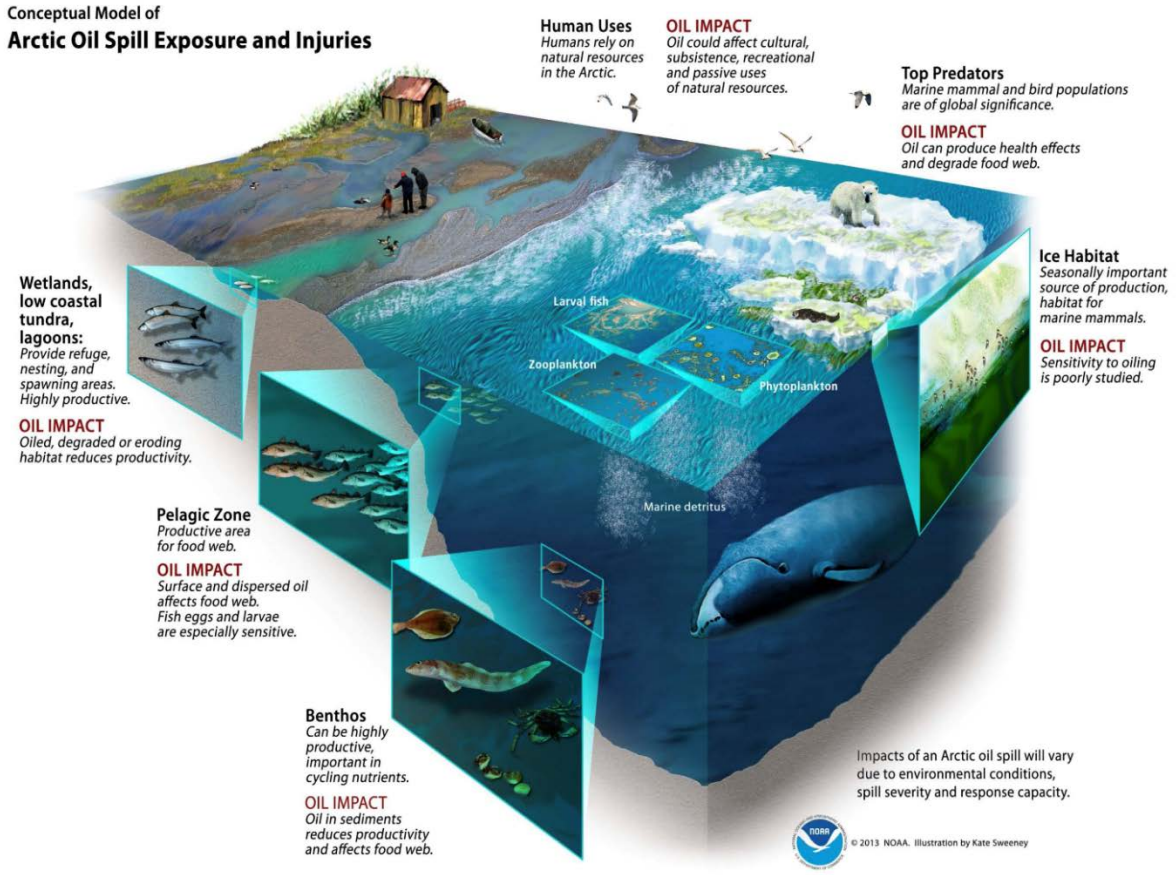


Figure 1. Conceptual model of Arctic oil spill exposure and injuries. Source: NOAA, 2013.

Research Planning, Inc. (RPI) and the National Oceanic and Atmospheric Administration (NOAA) Office of Response and Restoration, Assessment and Restoration Division developed sampling guidelines for collecting high priority ephemeral data in the event of an oil spill in the Arctic. These guidelines are detailed and include information specific to the area targeted for sampling. Guidelines have been developed for the following environmental media and biological resources:

Environmental Media

- Source oil
- Stranded oil on shorelines
- Oil sheen
- Water (surface and subsurface)
- Snow
- Intertidal and seasonally inundated lowland sediments
- Subtidal sediments
- Shellfish tissues

Habitats and Associated Communities

- Sand beach infauna
- Gravel beach communities
- Rocky intertidal communities
- Ice
- Vegetated habitat (marsh, low lands, tundra)
- Eelgrass
- Kelp-boulder fields

Resources

- Plankton
- Fish (ichthyoplankton, juveniles adults)

These guidelines will help ensure that ephemeral data collection methods used by NOAA, trustees, and others involved in the NRDA process, address the highest priority ephemeral data needs for injury assessment in the Arctic, collect data of sufficient quality and quantity to support NRDA, account for unique Arctic habitats, resources, and conditions, and are realistic in addressing the challenges faced by assessment teams when sampling in remote Arctic areas. Additional guidelines and other supporting information are also included in this document.

Important Definitions

- Ephemeral data are types of information that change rapidly over time and may be lost if not collected immediately (e.g., within days or weeks).
- For the purpose of these guidelines, the Arctic is defined as area encompassing the Bering, Chukchi, and Beaufort seas. The area north of the Bering Strait may be referred to as the 'high Arctic' when differentiating this region from the Bering Sea and Aleutian Islands.
- Sampling guidelines are general descriptions that can be used to facilitate ephemeral data collection when a sampling plan is not available. A sampling plan may provide additional details on "what, when, and how" samples and data will be collected. Consequently, a sampling plan is different from guidelines in terms of the level of detail and specificity.

Purpose

The purpose of these sampling guidelines is to provide general guidance on how to collect baseline and post-oiling information to assess the impacts of an oil spill in the Arctic on certain resources and habitats. These guidelines for collecting high priority ephemeral data will support NRDA and other oil spill science in the event of an oil spill in the Alaskan Arctic. It is important to obtain reliable biological and other environmental information that accounts for spatial and temporal variations of oil impacts. Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.

These guidelines are intended to be advisory and not mandatory. Spill-specific considerations, local field and weather conditions, personnel availability and training, logistical support, access restrictions and other factors may limit the ability to adhere fully to these guidelines. The adequacy of collected ephemeral data to guide relevant decisions should ultimately be judged by the circumstances of the spill involved.

Guideline Development Process

Draft ephemeral data collection guidelines were developed based on guidance documents for other regions, published sampling methods, lessons learned from other spills, meetings with communities in the Alaskan Arctic, and shared traditional ecological knowledge. Complete drafts of these guidelines were shared with Arctic resource experts and NRDA practitioners with expertise on subject-specific guidelines. Prior to their distribution, webinars were held to introduce the rationale and overall objectives of these guidelines to the subject-matter experts and likely end-users. Experts were asked to review, revise, and provide feedback on the guidelines to improve their quality and ensure that they are comprehensive enough to support high-quality ephemeral data collection. Key elements considered by the reviewers included:

- **Focus** – Guidelines should contain information on ephemeral data collection critical to providing information needed for injury assessment.

- **Completeness** – Guidelines should be as complete as practical to achieve NRDA goals in the event of an oil spill in the Arctic.
- **Articulated** – Guidelines should be as clear and accurate as practical to facilitate their use by technical and non-technical personnel.
- **Arctic-specific** – Guidelines should address, in as much detail as practical, considerations related to sampling in the Arctic.
- **Concise** – Guidelines should contain all the necessary information.
- **Attention to Critical Details** – Guidelines should contain QA/QC information to ensure that all the necessary supporting information is collected and recorded.
- **Adaptable** – Guideline documents should be flexible enough to be adapted to a variety of area- and spill-specific study plans.

Subject-matter experts and NRDA practitioners reviewed the draft guidelines in each of three categories: Media, Habitats, and Resources. All comments were reviewed and incorporated into revised guideline documents, which were sent back to those who provided comments for a second review. This was an important step to ensure that all comments or concerns were adequately addressed within the revised versions.

All of the environmental media and resource guidelines and four of the habitat guidelines were used in field validation exercises. Guidelines were tested at field sites in the high Arctic during the open-water season, including when broken ice was present along shorelines, to verify the practicality of implementing the guidelines as written, and to assess the level of effort required. Lessons learned from these validation exercises, as well as opportunities for improvement, were identified and addressed in the current versions of these documents.

Both the review by experts and field validation exercises were useful and helped make these documents practical and ensure that they address NRDA's need for high priority ephemeral data collection. All of the documents produced by this project can be updated and adapted as needed on an incident-specific basis. The goal is that they will be kept updated and ready to be implemented in the event of an oil spill in the Arctic.

Guideline Format

All guidelines have the same basic format and include field data forms. Sample collection procedures and field data sheets can be printed and taken into the field to guide sampling and for recording necessary information. All guidelines include:

- **Guideline objectives –** Describe guideline-specific objectives and provides other relevant information specific to each habitat or resource to be sampled.
- **Sampling objectives –** Describe the specific objectives that are to be achieved by collection of samples (e.g., study exposure, ensure collaboration).
- **Before field sampling –** Highlights important recommendations to be addressed prior to field sampling, including the importance of a more detailed sampling plan and a review of equipment and other preparations needed prior to field sampling.

- **Sampling areas and timing –** Includes a series of recommendations regarding selection of sampling areas, locations and sites and prioritization of sample collection that support the high quality data collection efforts required to meet the objectives of NRDA.
- **Sampling equipment/containers** – Provides a detailed list of equipment required to carry out the sampling described in the guideline.
- **Sampling strategy** – Provides detailed procedures and recommendations on how to collect samples and ensure their quality during collection, storage and transport. Specific aspects of this section include:
  - Quality assurance/quality control (QA/QC) information
  - Strategies for ensuring good sampling practices and decontamination
  - General guideline on the study design implementation
  - Details on sample collection methods
  - Guideline on sample labeling and record keeping, sample preservation, holding times and shipping
  - A list of analytical methods
- **References –** Provides a list of key references used during the development of these guidelines.
- **Appendix A –** Provides supporting documents including field data sheets.

In addition, all guidelines refer to three general NRDA guidelines developed to ensure the completeness of the sample collection process. These include:

- Guidelines for alternative sampling equipment and methods suggesting modifications to sampling methods if certain required equipment or facilities are not available in remote Arctic locations.
- Standard guidelines for Chain for Custody during sample storage, transportation, and shipping.
- Guidelines on how to take, document, transfer, and store photographs and video that support NRDA efforts.

Important Considerations

- These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples are taken.
- Each guideline document describes one approach, but other equally good options may be available for specific metrics or sampling methods.
- These guidelines should be adaptable and ready to be implemented during the first days and weeks of an oil spill in the Arctic by NRDA practitioners, in accordance with a sampling plan, if one is available.
- These guidelines highlight some of the most obvious scientific and logistical challenges of ephemeral sampling in the Arctic. These descriptions are not intended to be all-inclusive.
- Collaboration with other ongoing ephemeral sampling is important. Guidelines make specific reference to related guidelines, as well as other essential supporting documentation (e.g., Chain of Custody guidelines).
- These guidelines do not include:
  - Guidelines for bird exposure and injury surveys (developed by the U.S. Fish and Wildlife Service)
  - Guidelines for marine mammal exposure and injury surveys (under development by the NOAA National Marine Fisheries Service)

Key Assumptions Regarding Ephemeral Data Collection in the Arctic

During the development of these guideline documents, special attention was given to issues, challenges, and limitations associated with ephemeral data sampling in the Arctic. As a result, several specific assumptions are highlighted below:

Safety: It is assumed that all personnel working in the field have appropriate personal protective equipment and training. Safety consideration for Arctic field work, working with hazardous materials, and working on or operating vessels, aircraft and vehicles are not described in the sampling guidelines. The safety of personnel is the primary consideration when planning and carrying out sampling in the field.

Access to Supplies: It is assumed that all the essential supplies, equipment, and other specialized tools are accessible and available to the sampling teams. Therefore, it is important for those who are most likely to conduct these kinds of sampling to become familiar with the use of supplies and equipment, know where they can be obtained on short notice, and determine shipping options, weight restrictions, and shipping times to/from the region.

Access to Sampling Areas: It is assumed that all the logistical and safety challenges and consideration are carefully addressed before teams access sampling areas. While it is imperative that all personnel be aware of these challenges, other sections within the Unified Command may be responsible for ensuring that measures are in place to guarantee safe access to sampling areas. In addition, it is assumed that appropriate permissions for transit through or access to lands will be obtained before going into the field.

Working in the Arctic: It is assumed that any equipment going into the field in the Arctic and near water should be at least water-resistant and have battery life that will last in cold weather. All sampling teams should be aware of the fact that extremely cold temperatures shorten battery life, and therefore, sufficient batteries and/or backup power sources are needed when equipment that requires a power source are used for extended periods.

Working in the Dark: Some of these guidelines do not apply or have a restricted use during winter and most of the fall, primarily because of logistical and safety concerns for conducting field activities in the dark. When these challenges can be overcome, some of these guidelines can be implemented in the dark.

Working in the Presence of Ice: Some of these guidelines are not designed for use when shore-fast ice is present on shorelines, including intertidal habitats and sediments.

Technical Knowledge: Guidelines vary in the level of technical expertise required to meet the sampling objectives. While some methods may be carried out by personnel with basic scientific technical knowledge or minimal training, many require a high level of technical knowledge to be implement effectively. Some guidelines require close coordination with experienced personnel. In some instances, higher level of technical knowledge may be needed to assist with the implementation of specific methods. In addition, when time allows, a calibration exercise helps

ensure that all field teams perform sampling procedures consistently. Sampling teams must be composed of at least some personnel experienced in general and NRDA sampling.

Tidal Range: Guidelines often recommend running transects and conducting overflights within two hours of low tide (before or after). However, this recommendation varies depending on the geographical area to be sampled or surveyed. For example, tides may not be a consideration for shorelines along the Chukchi and Beaufort Seas where the tide range is small (<30 cm). In these areas, wind-driven storm surge may be the most determinant factor in water height and should be considered for sampling. Tides are mostly a consideration south of the Bering Strait where the tidal range is greater than 1 m.

Data Forms: In the absence of incident-specific data forms, sampling teams can use the forms provided within these guidelines, including Chain of Custody forms. However, incident-specific, sampling type-specific (e.g., habitat) and area-specific modifications may be needed to ensure the completeness and accuracy of the information collected.

Injury Documentation: While these guidelines are primarily intended to collect information used to characterize injuries from exposure to oil, ephemeral data collection can also be used to document impacts from cleanup activities and other response actions.

Injury Resulting from Sampling Activities: It is assumed that all sampling personnel are mindful of the potential impacts associated with conducting field studies on fragile and sensitive habitats and the potential for disturbance of wildlife. Field activities need to be conducted in a manner that reduces any potential impacts or disturbances to sensitive resources.

Coordination with Emergency Response: It is assumed that while an emergency response is ongoing, a NRDA liaison will coordinate with the Unified Command for the response as needed for information sharing, safety, logistics, resources, etc.

Interference with Response Actions: Ephemeral data collection and related sampling are not to interfere with response actions, including shoreline treatment.

Other Important Information

Arctic Safety Considerations

No samples or data are ever worth risking the health and safety of any team member!
Sampling in the Arctic can be extremely challenging (e.g., extreme weather conditions, remoteness and difficult access, dangerous wildlife, etc.) and special precautions must be taken. The safety of personnel is the primary consideration when planning and carrying out sampling in the field.
A daily go/no go decisions will be made for field activities, incorporating conservative safety assumptions.
All field personnel must be aware of all potential hazards and safety considerations, and they must have the necessary training to work safely and respond to emergency situations.
All field personnel must be aware of their surroundings and are responsible for compliance with all safety requirements.
Sampling in the Arctic along shorelines and near water in the dark during winter and most of the fall can be extremely hazardous, and extra safety precautions must be taken if sampling is required under these conditions. The use of an approved personal floatation device (PFD) is mandatory for each field team member.
Bear guards are required for on-shore field work in the Arctic.
Working with local people who know the area can improve the safety of field sampling activities.
All field personnel must have access to communication equipment, survival gear (appropriate to the conditions), and a first aid kit.
Careful logistic planning and support are critical to ensure the safety of sampling teams. Communication channels and methods, and drop off and pick up locations and times (e.g., by helicopter) need to be carefully considered and implemented.
Daily sampling activities must consider all Health and Safety issues and must ensure that all protective personal equipment is available prior to field sampling.
A detailed site safety plan must be in place and should include emergency response plans. All field team members must read and sign the site safety plan.

Data and Information Management

All data collected for NRDA will be part of a legal case, and proper data and information management is critical to the success of ephemeral sampling in support of NRDA efforts.

Engaging a data management team or expert in data and information management immediately after an incident is needed to provide essential guidance and QA/QC so that the data are useful from the time of collection.

Developing new field data forms or changing existing field data forms during the development of a new study plan should be coordinated with the data management team. Unauthorized changes to approved data forms are not allowed. The need for any changes to forms must be submitted to the data management coordinator, who will determine the need for modifications and make appropriate changes. Revised forms must be provided to all field teams before the next sampling mission.

It is important to that data forms are printed on waterproof paper and filled out with waterproof ink to ensure that water or moisture would not have an impact on the information collected.

Every person collecting ephemeral data must consistently and correctly complete data and chain of custody forms, as well as other forms of documentation. Training and familiarization with data forms are required before going in the field.

Every person collecting ephemeral data must follow the same instructions to complete data forms.

All data must forms must be complete, legible, consistent, and be both precise and accurate. All data fields must be filled out or labeled NA if not applicable.

Do not erase or black out erroneous entries on the field data forms or field notebooks. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through. If minimal space is available on the forms to indicate errors, use the back of the page indicating with arrows the location of the correct entry or an explanation of the erroneous entry. Additional notes can also be made on the field notebook, but a note on the data form must indicate that additional information resides in the field notebook.

It is important to review, correct, complete, sign, and submit all forms to the data management coordinator as soon as practical after field sampling. This includes all the data forms, field notes, photographs, videos, maps, and any other data collected in the field. When working in remote locations with limited connectivity, it is important to coordinate with the data management team.

Field data forms should be scanned and sent to the data management coordinator or a data management system if one has been established. If a scanner is not available, a digital photograph of the form can be used only if the text is legible. Field data forms that are filled out by hand in the field should be transcribed into a digital format, if one is available, and sent to the data management coordinator.

Once data have been collected from the field, coordinate with data management to upload data points to ERMA or other picture/mapping sources being used. This will help inform other ephemeral data collection plans and avoid overlapping efforts in ephemeral data collection.

Copies of all information types (data forms, field notes, photographs, videos, maps, and any other data collected in the field) must be made as soon as practical.

Digital versions of data forms specifically developed for ephemeral data collection in the Arctic. Coordinate data entry with NRDA data management personnel.

Sample Labeling

All guidelines for ephemeral data collection include details on sample labeling.
Sample identification and labeling is extremely important in the NRDA process, as it allows for tracking the individual samples throughout the case.
Some samples may be used immediately after collection, while others may be used in later analyses. It is important that all labels are secured to the sampling container, and that all precautions are taken to ensure that labels are clear, legible, accurate, secured to the container, and long-lasting.
If multiple teams are simultaneously collecting samples it is extremely important that a numerical coding scheme is planned ahead of time such that each sample is assigned a unique identification number.
Any changes to the sample numbering or labeling requirements should be coordinated by the data management team.
When labeling jars, record the sample number on both the label and lid. The sample number on the label must be complete and identical to the number entered on the Chain of Custody form. Jar labels should include a protective layer of clear tape wrapped around the entire circumference of the container to secure the label and protect the writing.
For samples placed in bags, place a sturdy waterproof paper label in indelible ink into the bag and repeat the label on the outside.
When and where possible, Sample Intake Centers should be used to ensure compliance and accurate sample labeling.

Hazardous Materials

All field personnel must be familiar with the chemicals to be handled in the field, and they must have access to Safety Data Sheets (SDS). These sheets include details on handling requirements, as well as all the essential protective personal equipment.
Hazardous materials regulations affect sample preservatives and other materials (e.g., formalin, alcohols, solvents, some cleaning agents etc.).
Hazardous materials can be shipped under some circumstances (e.g., samples containing ≤10% formaldehyde that do not exceed 30 mL per primary container or 1 L per shipment), but special packing is required (e.g., primary container, watertight secondary container, absorbent material between primary and secondary container, and sturdy outer packaging).
Hazardous materials may need to be shipped to the sampling locations via cargo or charter aircraft, and will need proper documentation and shipping containers to comply with transportation regulations.
Some of these shipping requirements may delay shipment of equipment to field study areas.
Health and Safety requirements need to be considered when handling hazardous materials in the field.

Overflights

Determining the extent of oiling or habitats and resources potentially in the path of an oil spill may require aerial overflights. Overflights require careful planning and coordination to meet the overall goals of these missions.
Given the remoteness of most areas within the Arctic, overflights may be limited or unfeasible. If overflights are feasible, these require experienced aerial documentation personnel to ensure that accurate and reliable information is collected.
Refer to the NOAA Office of Response and Restoration overflight safety procedures, or other relevant safety guidelines, for information about required training and emergency equipment for all personnel flying in helicopters or other aircraft over water.
At a minimum, OR&R personnel are required to be trained in helicopter underwater emergency egress and have proper emergency equipment including a personal emergency locator and SEA – small air supply bottle and specialized vest. Requirements for other personnel may be different.
Survival equipment applicable for the operating environment (e.g., personal flotation devices and survival suits) must be available to all personnel assigned to overflight missions.
An emergency locator device (e.g., Emergency Position Indicating Radiobeacon or EPIRB) must be present on the aircraft.
A safety briefing with the pilot and all crew members assigned to overflight missions must take place prior to boarding the aircraft. Safety briefings should include an overview of aircraft safety procedures, safety gear, radio operations, etc.
Prior to starting an overflight mission, learn the basic communications protocol (e.g., when to/not to talk, who is directing the pilot, and how to direct the pilot in terms of changes in elevation or distance offshore).
Prior to departing, revise the flight plan with the pilot. Have a map showing the flight plan.
Print copies of maps of the areas to be surveyed and plot the flight path on the map, noting the time every 30 minutes or when changing direction. Use of a GPS in track mode is recommended.
Keep track of your location and elevation at all times, in case of an emergency.
Make detailed observations of oiled natural resources or the proximity of natural resources (e.g., biological, habitat, etc.) and services (i.e., ecological and public) relative to the incident.
Take photographs and video, if practical, and ensure completeness of the photo log (see Field Photography guideline).
Identify the location of all photographs on the map as they are taken to create an accurate record of the overflight.
After completing an overflight mission, transfer all observations to a worksheet and a clean copy of a map.

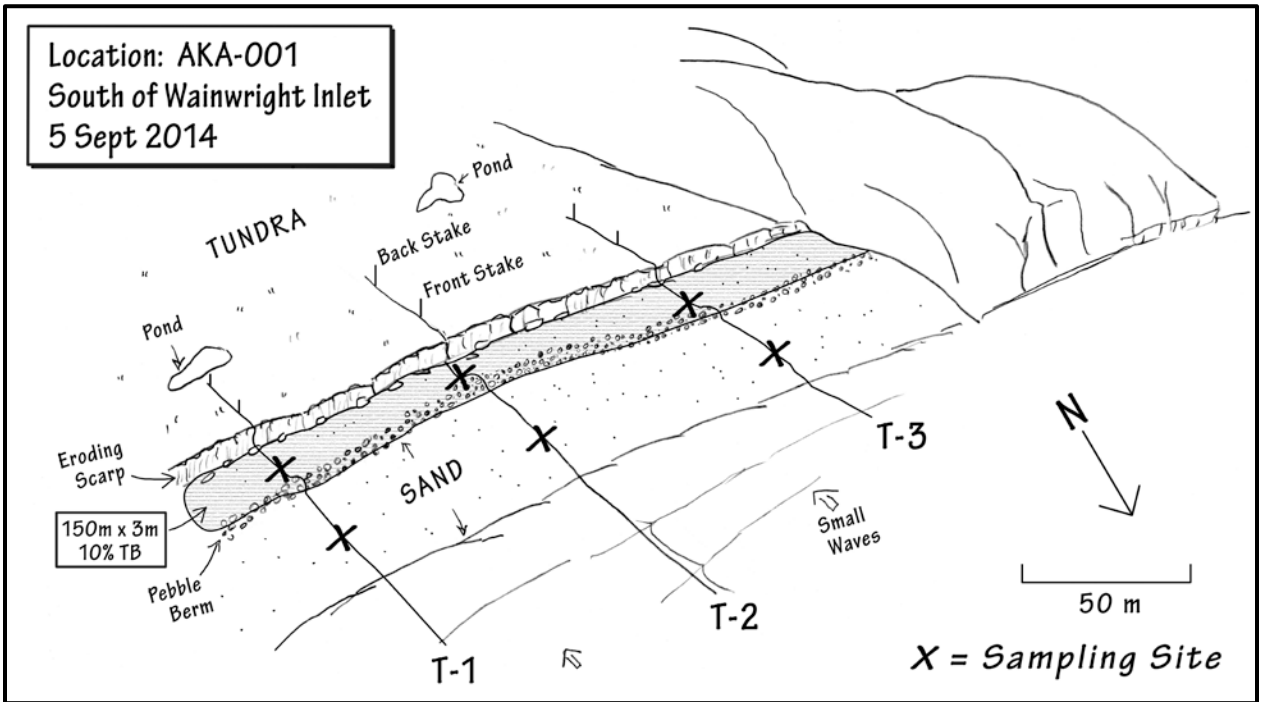
Sketching in the Field

Many of the guidelines suggest making a field sketch of the sampling site.

A field sketch is important because it can be used to re-occupy a site even if stakes or other markers have been lost. It also helps the person doing the sketch to make detailed mental notes of all the site features.

A field sketch can be either plan view or oblique. The figure below shows an oblique sketch of a sand beach site.

Be sure to include a scale, north arrow, location of the transects, sampling sites, grain size of the sediments, and the descriptions of any oil on the shoreline. Use a box to indicate the oiled band width, length, % oil cover, and oil descriptor (using standard SCAT terminology).



EPHEMERAL DATA COLLECTION GUIDELINES:

ENVIRONMENTAL MEDIA

Guidelines for Collecting Ephemeral Data in the Arctic: SOURCE OIL

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collection of source oil samples for chemical analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations.

Sampling Objectives

Characterize oil

- Obtain sample(s) of the original oil source(s) involved in the incident for characterization, fingerprinting, and predicting/quantifying oil fate and effects

Quality assurance/quality control

- Ensure the integrity of the spill source sample(s) throughout sampling, transport, and storage.
- Ensure the reliability of chemical characterizations

Collaboration

- Support other ongoing efforts (see Water, Snow, Ice, and Sheen guidelines)
- Provide material for toxicity testing to support NRDA

Before Field Sampling

- Access to the source oil may be restricted by emergency response operations. Because source oil collection is a potentially dangerous activity (e.g., spark risks, potential inhalation of volatile compounds, etc.) that requires highly technical training, emergency responders may collect source oil samples. Consequently, emergency responders could be provided information and documentation required for sampling to support NRDA. Coordinate with the emergency response before carrying out source oil sampling.
- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) may vary within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- Plan ahead the number of locations and samples to be collected, taking into account level of effort, potential logistical limitations, weather conditions, etc. that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.

- Contact the laboratories that will be receiving the source oil samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- The source oil sample should be collected from the freshest source possible (i.e., directly from the tank(s) of vessels, from the wellhead, or pipeline); secondary options are from the water or shoreline as close as practical to the spill source(s).
- If source oil samples are likely needed for post-spill laboratory studies (e.g., toxicity studies, etc.) collect as much source oil as practical (e.g., several liters up to a barrel). The team would be notified of these sampling needs by a NRDA supervisor.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Collaboration

- Source oil samples can be collected in conjunction with water, snow, ice, and sheen samples.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The type of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs− for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for source oil sampling under extreme cold conditions
- Sampling jars – certified organic-clean glass jars (solvent rinsed) with Teflon-lined lids and labels:
  - 1 L wide mouth glass jars, amber glass preferred
  - 250 mL wide mouth glass jars, amber glass preferred
- Samplers – Wheaton grab sampler, Volskom sampler (if available)
- Transferring sampler – drum thief, coliwasa, bailer, bomb sampler, or air-driven metallic pumps
- Stainless steel spatulas or spoons for collection of viscous oil samples
- Sampler material – Teflon (preferred), glass, or PVC (less ideal)
- Transferring containers (cleaned and solvent rinsed) – new metal/stainless steel (preferred), glass or plastic (less ideal)
- Funnels (for transferring; solvent rinsed) – metal/stainless steel (preferred), glass or plastic (less ideal)
- Bucket or a conical polyurethane bag (for source oil collection from surface water)
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- GPS, camera, field notebook, evidence tape (see Chain of Custody guidelines)
- Packaging materials for glass jars− may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional:

- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated

gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.

- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples:
  - Wash sampler and other equipment with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- Safety is of greatest concern. Be aware of physical and chemical hazards at the site. Get a safety briefing before entering the area. Do not enter confined spaces unless they have been determined to be safe. Use recommended safety equipment and procedures.
- Ship-source sampling is a highly technical and potentially dangerous activity that should ideally be undertaken by vessel crew, marine surveyors, or salvors. Consider requesting source oil samples through the emergency response unified command.
- There are spark risks when sampling oil and sampling may be restricted to specific areas of a vessel and subject to strict supervision.
- There may be different products loaded in different compartments or tanks, even when only one type of product is involved (e.g., two different types of No. 2 fuel oil could be loaded). Before sample collection, get a loading diagram and as much data as practical on the products. Slight differences in product properties, such as specific gravity, may indicate different products. Each unique product should be sampled.
- Make sure that the source sample(s) collected represent the material actually released. Follow any information/evidence from crew and other personnel to ensure that source samples are collected from the released material. Again, product in intact holds may not be the same as the released product; however if feasible, it may be appropriate to collect samples of other products of concern in case of

future releases. Samples should be collected from compartments or tanks that are compromised, or that are at risk of becoming compromised.

- Collect the source sample as soon as practical, even for potential releases. Collect the freshest sample of the spilled material, even if it is from the water, shoreline, etc.
- Collect multiple samples in case samples are compromised, or when multiple locations are to be sampled (e.g., per tank, hold, etc.). Preferably, collect 1 L triplicate samples per source; a minimum of 1 L per source.
- If sampling source oil through a spigot, purge the initial flow into a designated oil recovery container and collect the source oil sample after a few seconds of flow directly into the sample container.
- When collecting source oil(s):
  - Source samples can be collected directly into the sample container, minimizing risks of contamination. Use a device that holds the container, such as a Wheaton grab sampler (holds a 1 L bottle strapped to a metal rod with the ability to unscrew the cap remotely) or a Volskom sampler (frame which holds a container lowered on a rope). For black oils, these methods are less ideal because of coating of the device and container
  - Samplers for transferring source oil from a hold to a container include drum thief, coliwasa (narrow tube with positive end seal), bailer, bomb sampler, and air-driven metallic pumps
  - Sampler material, in order of preference, include Teflon (there are inexpensive disposable models), glass, and PVC (may contribute plasticizer chemicals such as phthalates, but should not interfere with interpretation)
  - Under emergency conditions use a glass container or a new metal/stainless steel (preferred) or plastic (less ideal) bucket to collect the source oil by scooping or dipping, etc.
  - Pour contents or use funnel to transfer into another container so the outside of the transport container is kept clean, reducing the chance of cross contamination
  - Leave approximately 1 cm headspace in the neck of each jar. A larger headspace (e.g., 2.5 cm) is recommended to allow for sample expansion if water is present in the sample and freezing may occur
  - Care should be taken not to contaminate the outsides of the lid or the sample jars. If contamination does occur, the containers should be cleaned with soap and water, or less preferably with sorbent material. Do not store containers that have external oil contamination with other samples until they have been cleaned
- On occasion large samples are not available. Any size representative sample of the source oil is better than no sample at all. As a last resort, collect 250 mL (3 replicates if possible) of source oil from each hold or tank. If the collection of samples directly from the source is not possible, 1 L samples of oil should be ideally collected in triplicates from the water or shoreline as close to the source as practical.
  - When collecting source oil from water surface, concentrate the source oil in the sample container by skimming the oil from the water, sampling from the thickest part of the freshest oil slick. If using a bucket or a conical polyurethane bag with small holes at the bottom, skim oil and allow water to drain before transferring contents to the sampling jar
  - When collecting source oil from the shoreline, carefully scrape the thickest part of the stranded source oil from the shoreline surface into the sample container minimizing the transfer of sediment or any other foreign material
- Be aware of sources of contamination or mixing of products on-scene, such as transferring of product between holds, dilution with fire-fighting water, or application of a foam blanket. If contamination is suspected, it may be appropriate to take samples of the potential source of contaminant.
- After each separate likely source tank, hold, or location is sampled, decontaminate non-disposable equipment (described earlier).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.

- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each oil sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make notation on the Chain of Custody form about any problems or observations during sampling, such as visual differences in samples from different tanks, presence of water in the sample, etc.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record on the data sheet:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample #, date/time
  - Sample matrix (source oil)
  - Indicate if samples were collected at the source, or from nearby water or shorelines
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately following collection, place all source oil samples in cooler and keep at 4°C. DO NOT FREEZE. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature if heated storage space is not available. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Protect the samples from direct sun exposure (e.g., UV radiation).
- Tape lids on sample bottles so that they do not accidentally come off.
- Refrigeration temperature shall be recorded upon sample storage; monitor and record refrigeration temperature periodically to ensure proper refrigeration.
- Use packing material, such as bubble wrap, around containers to prevent breakage during handling and shipping.
- Ship source oil samples separately from environmental samples to reduce risk of cross contamination and include complete Chain of Custody forms.
- Ship to the laboratory as soon as practical with completed Chain of Custody forms.
- Do not discard ANY source oil samples.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels	Recommended Holding Time[a]	Minimum No. of Samples per Oil Source
Various depending on the petroleum product tested	1 L per oil source [b, c, d] (preferred) 250 mL per oil source (minimum)	Various depending on the petroleum product tested	Indefinitely [e]	3 (preferred) 1 (minimum)

[a] Store at 4°C in the dark; [b] Several analyses can be made from a single sample; [c] Larger volumes (e.g., several liters up to a barrel) may be needed for post-spill studies (e.g., toxicity studies, etc.); [d] collected at the source (preferred), or from water or shorelines as close to the source as practical (less ideal); [e] If held at 4°C in the dark and without organic material or water, which promote bacterial growth.

Analytical Methods

- **Petroleum products** are usually analyzed for fingerprinting purposes, so that the spilled oil can be differentiated from other petroleum sources. Chemical analyses for this purpose include:
  - A quantification of the organic constituents of an oil sample is commonly determined by capillary column gas chromatography (GC) coupled with Flame Ionization Detection (FID) or for the most accurate and detailed analysis, Mass spectrometry (MS). The term "biomarkers" is also commonly used in fingerprinting and refers to the calculation of peak area ratios for a range of the more stable hydrocarbon compounds including hopanes, isopreniods and steranes
  - **Saturated hydrocarbons** (SHC). These compounds include n-alkanes and saturated isoprenoids. They are contained in the *f1* fraction obtained after column chromatography and measured by GC-FID analysis. The chromatographic trace can be used to differentiate among oils and is valuable for predicting the short-term weathering and fate of the oil
  - **Polynuclear aromatic hydrocarbons** (PAH). PAHs are used to fingerprint the spilled oil, monitor weathering, and predict toxicity. If PAHs are to be measured, it is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard PAH "priority pollutants." This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs (approximately 43 PAHs), using GC/MS in the selected ion monitoring (SIM) mode
  - **Petroleum biomarkers**: These compounds are highly resistant to degradation and have a unique distribution for each oil type. Thus, they are valuable for differentiating among different sources of hydrocarbons. However, few laboratories have the capability for quantitative analysis of biomarkers, which is a specialized method using GC/MS in the SIM mode
  - **Other characterization analyses may include**: density, boiling point curve, metal and sulfur content, weight fraction of aromatics, total paraffins, asphaltenes/resins, and sulfur in oil

Key References

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NOAA. 1993. Sampling and analytical methods of the National Status and Trends Program, National Benthic Surveillance and Mussel Water Projects, 1984-1992. Volume IV, Comprehensive descriptions of trace organic analytical methods. Lauenstein, G.G. and A.Y. Cantillo (eds.). NOAA Tech. Memo NOS ORCA 71, Silver Spring, MD. 181 pp.

Sauer, T.C. and P.D. Boehm. 1995. Hydrocarbon chemistry for analytical methods for oil spill assessments. Marine Spill Response Corporation Technical Report Series 95-032, Washington, D.C. 114 pp.

USEPA. 1979. Methods for chemical analysis of water and wastes. EPA-600/4-79/020. USEPA Environmental Monitoring Systems Laboratory, Office of Research and Development, Cincinnati, Ohio.

USEPA. 1986. Test methods for evaluating solid waste. SW 846 Third Edition (and updates).

Wang, Z. and S.A. Stout. 2006. Oil Spill Environmental Forensics: Fingerprinting and Source Identification. Elsevier Publishing Co., Boston, MA. 554 pp.

Appendix A
Supporting Documentation- Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Oil/Tarball/Water/Snow/Ice/Sheen Sample Collection Form

Sample Collection Form - OIL/TARBALL/WATER/SNOW/ICE/SHEEN										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling Method**	**Sample Position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(O)il, Tarball (B), (W)ater or Other (H)*	*Sample # (Team ID – sequential number) and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Method of sampling (i.e., grab or other)*	*Collection depth of water sample. Use 0 for surface samples.*	*Volume of sample with units*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD -YYY.YYYYYY*	*Description of sample, equipment used, photo numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										

Samples Relinquished by:				**Received by:**			
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball, Water, Snow, Ice, Sheen	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H) (SN)ow (I)ce (SH)een	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Complete sample IDs are comprised of the following information: Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Guidelines for Collecting Ephemeral Data in the Arctic: STRANDED OIL SURVEYS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which surveys may be conducted. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on characterizing and quantifying oil stranded on shoreline habitats during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. This document focuses on surveys rather than sample collection, which is addressed within in other guidelines.

Survey Objectives

Characterize shoreline oiling

- Quantify the amount of stranded oil on shoreline habitat for the following applications:
  - Support a mass balance for the fate of the spilled oil
  - Classify the shoreline segment into oil exposure categories (e.g., heavy, moderate, light) in order to illustrate oil distribution patterns
- Supplement Shoreline Cleanup Assessment Technique (SCAT) surveys conducted by responders, or to obtain more detailed oiling information at certain sites

Collaboration

- Support other assessment efforts including, but not limited to, assessment of sediment contamination. Oil samples should be collected to in conjunction with shorelines surveys to determine the source of the oil and characterize weathering state (see relevant shoreline habitat guidelines, e.g., Intertidal Sediment and Gravel guidelines, and the Source Oil guidelines).

Before Field Surveying

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) may vary within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the surveying team.

Study design

- It is important to have a defined surveying strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific surveying geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made

- Plan ahead the number of locations to be surveyed, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the surveying guideline may be needed based on environmental conditions, geography, and access to remote areas.
- The surveying strategy should have flexibility to be adjusted based on conditions in the field.
- Consider collecting stranded oil samples in conjunction with the survey if the source of the oil needs to be verified or determined. A small number of samples from an oiled location can be used to determine source even when other analyses are not planned (see Intertidal Sediment guideline).
- Use tools such as ShoreZone imagery, satellite imagery, aerial photographs, maps and Environmental Sensitivity Index (ESI) maps to plan surveys. Shoreline segments can be defined before going into the field.

Equipment
- Review the list of surveying equipment, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.

Survey Areas and Timing

- Plan all surveying strategies within daylight hours; surveying in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- Shoreline surveys may not be feasible if significant snowfall has covered stranded oil.

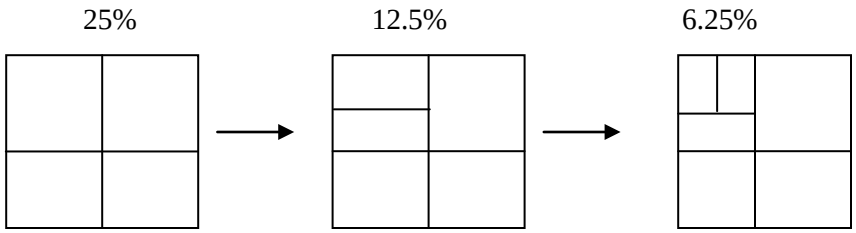
Collaboration
- Support other assessment efforts (see Source Oil, Intertidal Sediment, Gravel guidelines).

Field Surveying Methods

Survey Equipment/Guides
Note: The type of equipment required depends on the sampling plan, desired sample volumes, and logistics. If samples are to be collected during oil surveys, analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field (see appropriate guideline).
- Survey segment maps
- Tape measure (30 m)
- Shovel
- Small ruler for thickness measurements
- SCAT forms, terminology code sheet
- Percent cover estimator guides
- Camera, photo scale
- GPS
- Clipboard, pencils, field notebook
- Calculator (one per person)
- Roller counter

Calibration Exercises
- Conduct group calibration/training exercises with all survey team members prior to field surveys.

- To calibrate "Oil Distribution" estimates (% cover), use a quadrat to estimate the percent cover, or the following method. Draw a ~1 m^2 box in an oiled zone. Have everyone estimate % oil cover. Mentally herd the oil into one corner or, if possible, actually move the oil into one corner of the box. Draw a line to divide the original box in half. Keep halving the area until all of the oil fits into a corner. Estimate the area of the oil and compare with the original estimates. Repeat for different amounts of oil, and until everyone is making similar estimates. See diagram below:

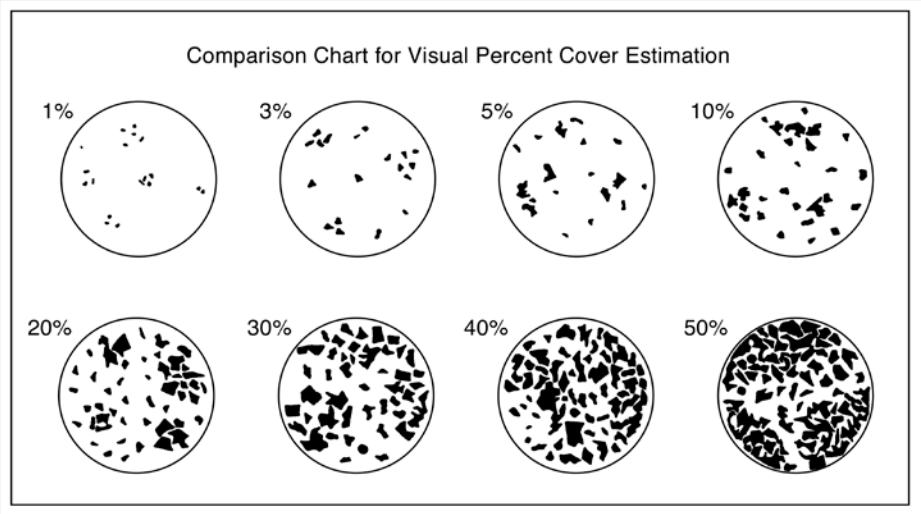


- To calibrate Length and Width estimates, use one of the following:
  - Measure out 30 m with a tape and have everyone determine their pace
  - Select various distances for visual estimation, then actually measure distances with a tape
  - If using a roller counter, calibrate it for the actual substrate using a tape
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Photograph the segment, zones, and pits to document oiling conditions. Make sure each photograph or series can be later associated with the corresponding surveyed segment, zones, or pits (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).

Survey Methods

- Use SCAT forms and standard terminology.
- Assign a unique **Segment Name** to the shoreline that you are surveying. Pre-assign segment names to prevent duplication. Coordinate with response staff and use SCAT segments if available. A **Segment** is a section of shoreline for which oiling observations are recorded. It is generally 0.2 to 2 km in length and has relatively similar geomorphology and oiling conditions. Each segment should be readily identified in the field (e.g., between two headlands, borders defined by streams).
- Walk or view the entire segment to get a sense of the different oil zones. Use a different **Zone ID** for each different oil occurrence, e.g., two distinct bands of oil, one at the high-tide line and one at a higher storm line, or along shore where the oil distribution changes from 10% to 50%.
- Within each shoreline segment collect information on:
  - Segment Name, date, time, weather conditions (e.g., wind direction and speed), tide level, and names of observers
  - Physical setting (shoreline orientation, exposure to wave energy and tidal currents, wind, potential for burial by sediment accumulation, sediment characteristics)
  - Shoreline habitat type
  - Dominant species or types of biota present
  - Presence of ice or snow
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
- Describe <u>each different</u> zone of **SURFACE** oil (defined as oil on the surface and penetrated up to 5 cm below the surface) in terms of its:

- **Zone ID**: for consistency, use A for the first one described, then B, C, etc.
- **ESI Type:** record the shoreline type using the ESI codes
- **Start/End Waypoints**: use a GPS and record the beginning and end of each segment
- **Tidal Zone Location**: the location relative to low, mid, high, or supra (above high) tide
- **Width**: the average distance of the oil zone perpendicular to the shoreline
- **Length**: the distance of the oil zone parallel to (alongshore) the shoreline
- **Oil Cover**: the average percent cover of the oil in the zone (to the nearest 5%) (see chart below)
- **Oil Thickness**: use SCAT terms (stain, coat, cover, etc.) or record thickness in cm to describe the thickness of any free oil on the surface; if >1 cm, record the actual thickness in cm
- **Oil Character**: use standard SCAT terminology (fresh oil, mousse, tarballs, patties, etc.)
- **Sediment Oiling Interval**: where surface oil has penetrated into the sediment, record the thickness of the oiled layer, from the sediment surface to the bottom of the oil (e.g., 0-3 cm) in the comments section
- **Samples Collected**: Collect representative samples (e.g., tarballs, intertidal sediments) of each type of surface oiling from several different segments so that the actual oil content can be measured by chemical analysis and used to calculate total oil volumes. Refer to the Intertidal Sediment guideline
- **Photograph**: Use a photo scale to take representative photographs of each oiled zone



- Determine the presence and extent **SUBSURFACE** oil (defined as oil penetration into the sediments >5 cm or buried by any thickness of clean sediments; see photograph). Assess the presence of subsurface oil by digging pits along the shore in areas of likely burial, such as high-tide berms, depositional areas near groins, or any subtle mounds of sediment that suggest recent accumulation or deposition of sediments. If subsurface oil is found, dig additional pits to define the areal extent of the subsurface oil. For each area of subsurface oil, record the following information:
  - **Pit #**: assigned as the oil zone and the pit number in that zone, e.g. A-1, B-1, B-2
  - **Location**: use a GPS and record the waypoint of each pit
  - **Substrate Type**: record the substrate type

(cobbles, pebbles, sand, mud, snow, ice, etc.) on the surface and in the subsurface
  - **Tidal Zone Location**: the pit location relative to low, mid, high, or supra (above high) tide
  - **Pit Depth**: the total depth of the pit (see photograph of a typical pit)
  - **Oiled Interval**: for each pit, record depths (from the sediment surface) to the top and bottom of the subsurface oiled layer (e.g., 5-10 cm, indicating that the subsurface oil layer started at 5 cm below the surface and was 5 cm thick). For multiple oil layers, record the interval of each layer
  - **Subsurface Oil Character**: describe the degree of the oiled sediment using SCAT terminology (oil-filled pores, partially filled pores, oil residue, stain, film, etc.)
  - **Samples Collected**: collect representative samples of each type of subsurface oil from several different segments so that the oil content can be measured by chemical analysis and used to calculate total oil volumes. Refer to the Intertidal Sediment guideline
  - **Photograph**: use a photo scale to take a photograph of every pit
  - **Estimate the Area of Subsurface Oil**: using the pit data, estimate the length and width of subsurface oil and record it in the comments section or a field notebook. Keep a detailed photo log so that each photo can be labeled and located as to the oil zone it represents. Time-synchronized GPS waypoints are a valuable tool when assigning coordinates to numerous photographs
- Make a sketch of the entire segment you just surveyed, showing the locations of all oil zones and pits.
- Make sure your notes are complete and include the segment name, date and time of survey, tidal level at the time of the survey, unusual weather conditions, and team member names/affiliations.
- Samples may be needed for chemical fingerprinting analysis or monitoring of weathering, to correlate a visual description of sediment oiling degree with actual oil concentration, or to confirm the absence of oil. Be aware of the potential for secondary contamination of the site from oil on boots and shovels when collecting samples. Refer to the Intertidal Sediment guideline.

Analytical Methods

- Refer to those under Intertidal Sediment guideline, if applicable.

Key Reference

NOAA. 2013. Shoreline Assessment Manual, 4th Edition. Emergency Response Division, Office of Response and Restoration, National Oceanic and Atmospheric Administration. 73 pp + appendices.

Appendix A
Supporting Documentation - Field Data Forms

The NOAA Shoreline Assessment Manual can be downloaded from:
http://response.restoration.noaa.gov/sites/default/files/manual_shore_assess_aug2013.pdf.
Note: These guidelines are not Arctic specific and site specific modifications and considerations are recommended.

Each member of the assessment team should have a job aid. Laminated shoreline assessment job aids can be ordered from NOAA for $20. Digital copies of the job aids are available from:
http://response.restoration.noaa.gov/sites/default/files/jobaid_shore_assess_aug2007.pdf

Additional shoreline assessment forms for wetlands and tar balls are available at
www.response.restoration.noaa.gov

Unique field data forms may be available; if not, use the SCAT Shoreline Oil Summary (SOS) form:

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes in the comments section of the SOS form. Label field notes appropriately so additional notes can be linked back to the corresponding SOS form.
- Fill in blanks with "N/A" if data is not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Coordinate data entry with NRDA data management personnel.

Attached form:
- SCAT Shoreline Oil Summary (SOS) form (with instructions and definitions of terms) and sketch map

SHORELINE OIL SUMMARY (SOS) FORM: _____ **Spill** **Page**____ **of** _____

1. GENERAL INFORMATION	Date (dd/Month/yyyy) (please use month name)	Time (24h standard/daylight) (00:00 to 00:00)	Tide Height L / M / H Rising / Falling
Segment ID:			
Segment Name:		____:____ to____:____	
Survey By: Foot /ATV/ Boat / Helicopter / Overlook / Other _____		Weather: Sun / Clouds / Fog / Rain / Snow / Windy / Calm	

2. SURVEY TEAM	Name	Organization	Name	Organization
Team Number				

3. SEGMENT	Total Length: m	Length Surveyed: m	Datum: WGS84
Survey Start GPS:	WP:	LAT: .	LONG: .
Survey End GPS:	WP:	LAT: .	LONG: .

4a. BACKSHORE CHARACTER: Indicate only ONE Primary type and ALL Secondary types

Cliff/Slope___ Lowland ___ Beach____ Dune___ Wetland___ Lagoon___ Delta___ Channel___ Man-Made____:_____

4b. ESI SHORELINE TYPE: Indicate only ONE Primary (P) and ANY Secondary (S) types. CIRCLE those oiled.

Primary:	Secondary:

5. OPERATIONAL FEATURES	Oiled Debris? Yes / No	Type:	Amount: (bags)
Direct backshore access? Yes / No	Alongshore access from next segment? Yes / No	Suitable for backshore staging? Yes / No	
Access Description / Restrictions:			

6. OILING DESCRIPTION: Indicate overlapping zones in different tidal zones by numbering them (e.g. A1, A2)

Zone ID	ESI Type	WP Start	WP End	Tidal Zone				Oil Cover						Oil Thickness					Oil Character							
								Area		Distribution		Size														
				LI	MI	UI	SU	Length (m)	Width (m)	Distr % >1	# per unit area	Avg Size (cm)	Large Size (cm)	TO	CV	CT	ST	FL	FR	MS	TB	PT	TC	SR	AP	No

7. SUBSURFACE OILING CONDITIONS: Format: Zone ID dash Trench Number in that Zone, e.g., "A-1, B-1, B-2"

Pit #	WP	Substrate Type Surface / Subsurface	Tidal Zone				Pit Depth (cm)	Oiled Interval (cm-cm)	Subsurface Oil Character										Water Table (cm)	Sheen Color B,R,S,N	Clean Below Yes / No
			LI	MI	UI	SU			OP	PP	OR	OF	TR	TB	SR	AP	NO	%			

8. COMMENTS: Cleanup Recommendations; Ecological/Recreational/Cultural Issues; Wildlife Observations; Oiling Descriptions

Sketch: Yes / No Photos: Yes / No Photo Numbers: (-) Photographer Name:

SHORELINE OIL Summary Form Instructions

Calibration IS VERY IMPORTANT! Do a calibration exercise to make sure that all teams are consistently using the same terminology and estimations.

Tide Height: Circle the letter indicating the tidal stage during the survey, and if the tide was rising or falling.

Segment/Survey Length: Always record both segment and survey lengths on the first survey, especially where the SCAT team creates the segments in the field. On repeat surveys, always enter in the Survey Length, especially if only part of the segment is surveyed.

Start/End GPS: The preferred format for latitude and longitude is decimal degrees, but be consistent among teams. Record the datum if different than WGS84.

SURFACE OILING CONDITIONS

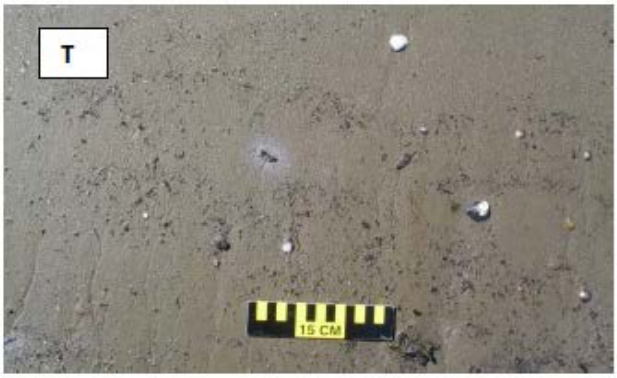
Zone ID: Use a different ID for each oil occurrence, e.g., two distinct bands of oil at mid-tide and high-tide levels, or alongshore where the oil distribution changes from 10 % to 50%. Describe each oil occurrence on a separate line. Record the shoreline type(s) present in each oiled zone using the terminology in section 4 or the ESI code (see box below).

ESI No.	Shoreline Type	ESI No.	Shoreline Type
1A	Exposed rocky shores	8A	Sheltered rocky shores
1B	Exposed, solid man-made structures	8B	Sheltered, solid man-made structures
2A	Exposed wave-cut platforms in bedrock, mud, or clay	8D	Sheltered rocky rubble shores
3A	Fine- to medium-grained sand beaches	8E	Peat shorelines
3C	Tundra cliffs	9A	Sheltered tidal flats
4	Coarse-grained sand beaches	10A	Salt- and brackish-water marshes
5	Mixed sand and gravel beaches	10B	Freshwater marshes
6A	Gravel beaches	10C	Swamps
6B	Riprap	10D	Scrub-shrub wetlands
7	Exposed tidal flats	10E	Inundated low-lying tundra

Tidal Zone: Use the codes to indicate the location of the oil being described, as in the lower (LI), mid (MI), or upper (UI) intertidal zone, or in the supra (SU) tidal zone (above the normal high tide level).

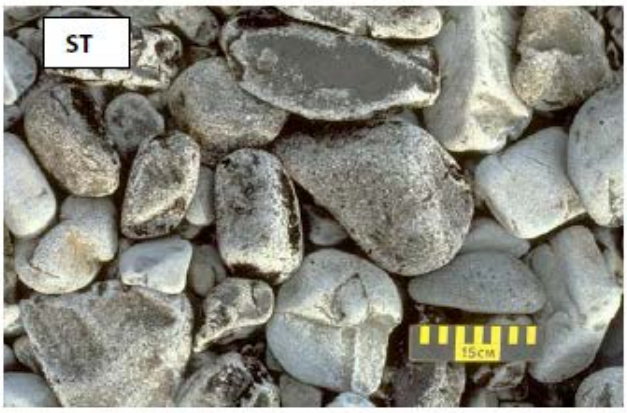
Distribution: Enter the estimated percent of oil on the surface (preferred), or codes for the following intervals:

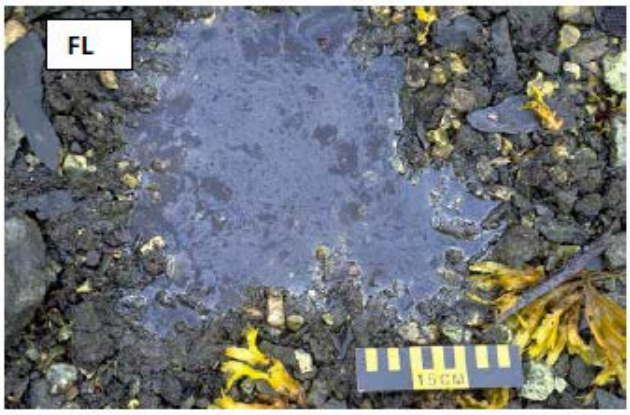
C	Continuous 91-100% cover
B	Broken 51-90%
P	Patchy 11-50%
S	Sporadic 1-10%
T	Trace <1%



Surface Oiling Descriptors - Thickness: Use the following codes:

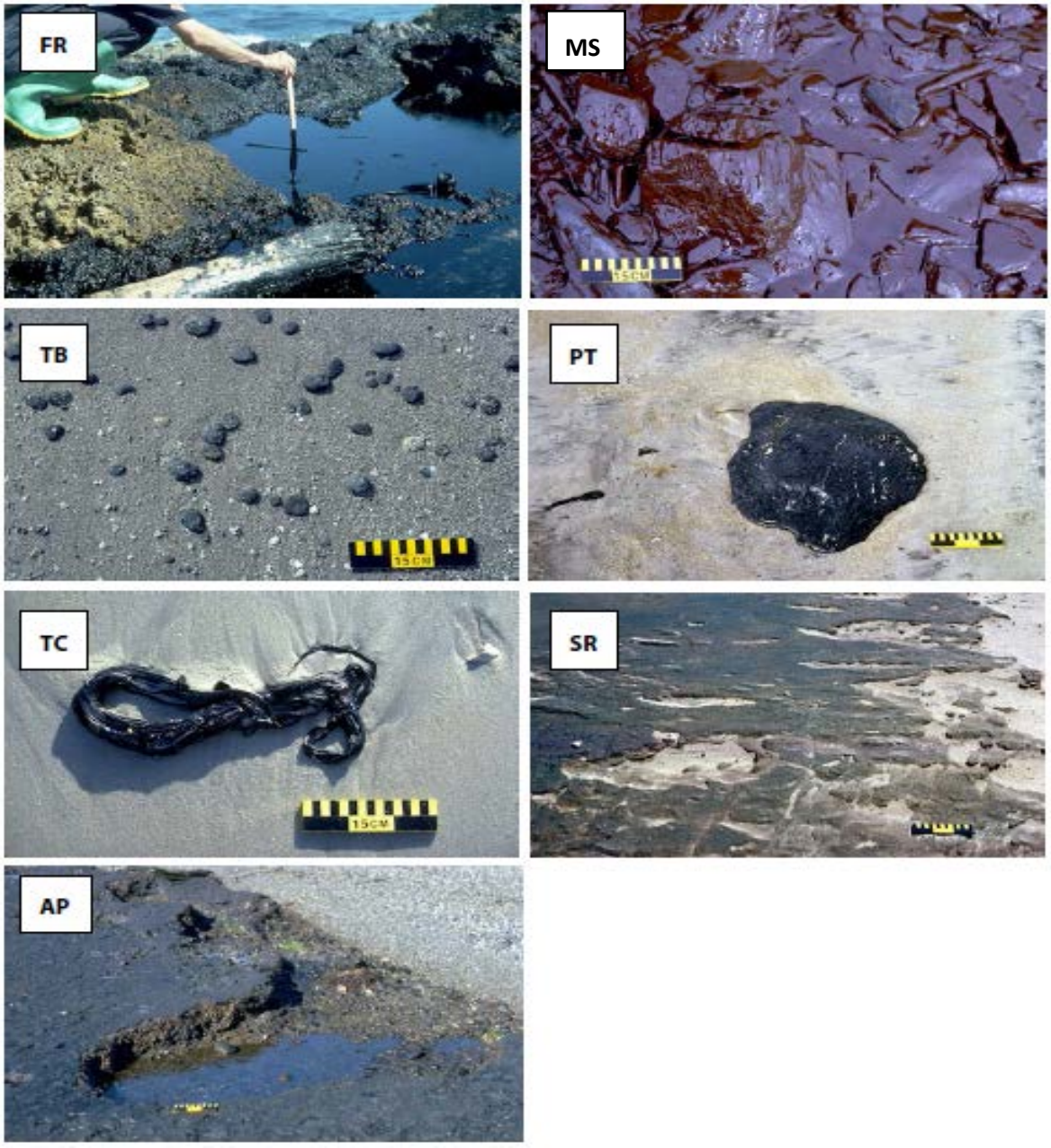
TO	Thick Oil (fresh oil or mousse >1 cm thick)
CV	Cover (oil or mousse from >0.1 cm to <1 cm on any surface)
CT	Coat (visible oil <0.1 cm, which can be scraped off with fingernail)
ST	Stain (visible oil, which cannot be scraped off with fingernail)
FL	Film (transparent or iridescent sheen or oily film)





Surface Oiling Descriptors - Type

FR	Fresh Oil (unweathered, liquid oil)
MS	Mousse (emulsified oil occurring over broad areas)
TB	Tar balls (discrete accumulations of oil <10 cm in diameter)
PT	Patties (discrete accumulations of oil >10 cm in diameter)
TC	Tar (highly weathered oil, of tarry, nearly solid consistency)
SR	Surface Oil Residue (non-cohesive, oiled surface sediments)
AP	Asphalt Pavements (cohesive, heavily oiled surface sediments)
NO	No oil (no evidence of any type of oil)

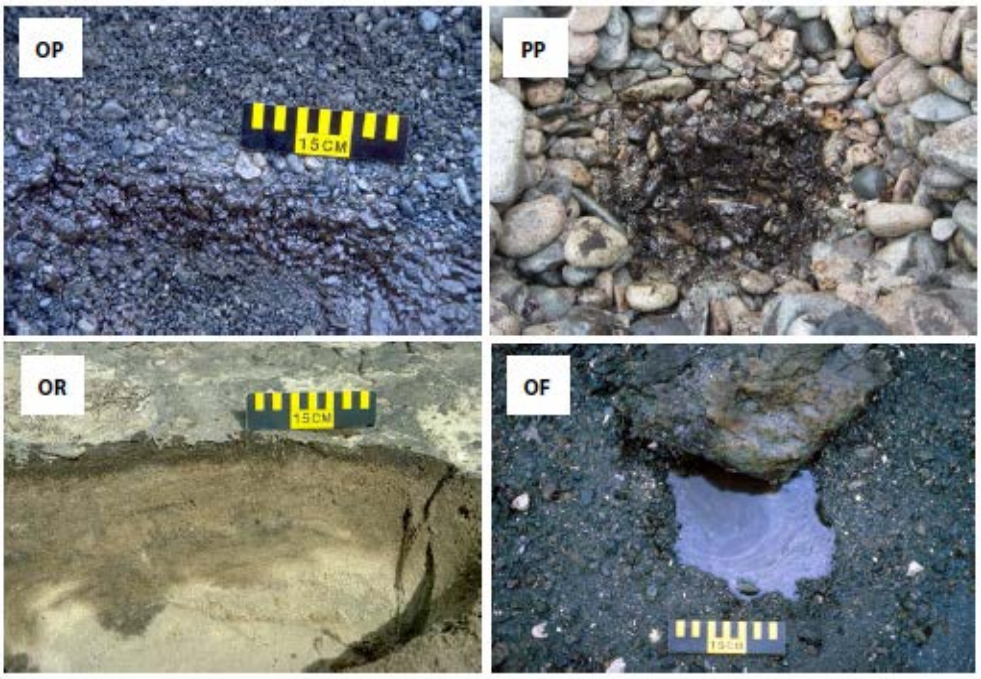


SUBSURFACE OILING CONDITIONS

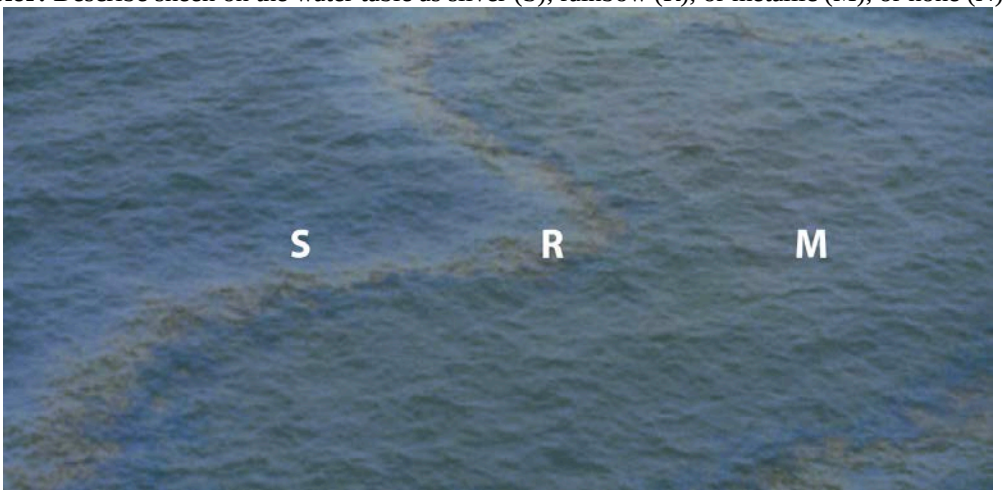
Oiled Interval: Measure the depths from the sediment surface to top/bottom of subsurface oiled layer. Enter multiple oil layers on separate lines.

Subsurface Oiling Descriptors: Use the following codes:

OP	Oil-Filled Pores (pore spaces are completely filled with oil)
PP	Partially Filled Pores (the oil does not flow out of the sediments when disturbed)
OR	Oil Residue (sediments are visibly oiled with black/brown coat or cover on the clasts, but little or no accumulation of oil within the pore spaces)
OF	Oil Film (sediments are lightly oiled with an oil film, or stain on the clasts)
TR	Trace (discontinuous film or spots of oil, or an odor or tackiness)



Sheen Color: Describe sheen on the water table as silver (S), rainbow (R), or metallic (M), or none (N).



SKETCH MAP

Site Name _____
Site No. _____
Date _____
Time _____
Names_____
_____

Checklist
___North Arrow
___Scale
___Oil Distribution
___High Water Line
___Low Water Line
___Substrate Types
___Trench Locations

Legend

#△
Trench Number
No Subsurface Oil

#▲
Trench Number
Subsurface Oil

#●→

#●

Photographs

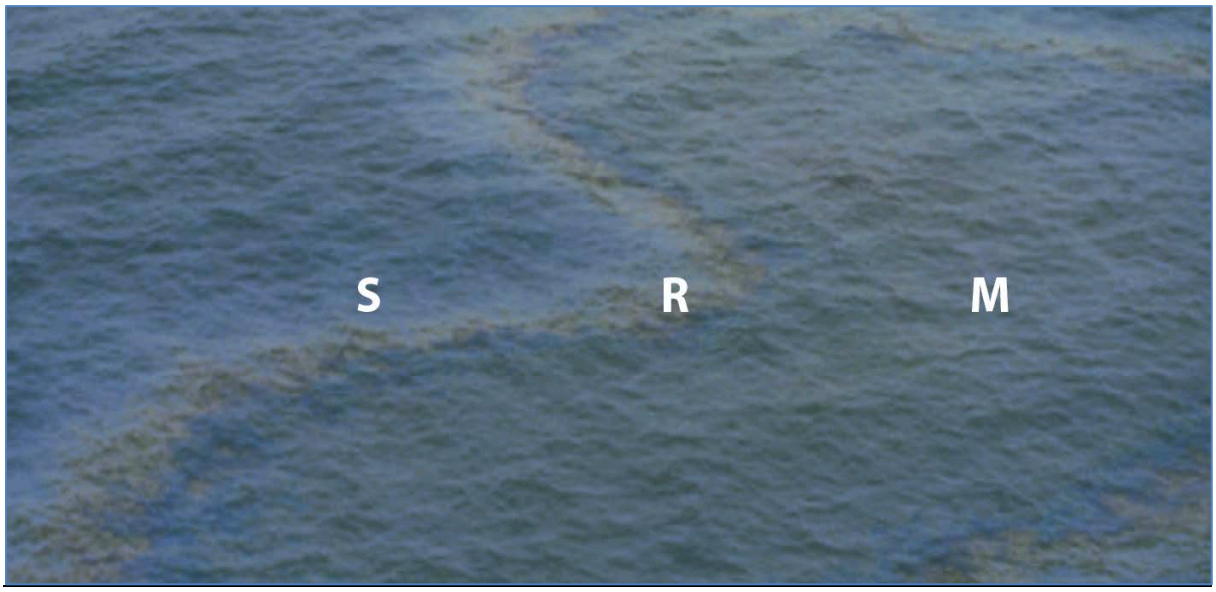
Guidelines for Collecting Ephemeral Data in the Arctic: SHEEN

July 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collection of sheen samples for chemical analysis from the water surface during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. By definition, sheen is a very thin layer of oil on the water surface that is described as silver (S), rainbow (R), or metallic (M) as shown in the image below.



Sampling Objectives

Characterize oil

- Document the presence of oil, and characterize oil weathering and fate
- Determine the source of contamination via chemical fingerprinting analysis
- Document the presence of oil, and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Study exposure

- Document exposure of water-surface organisms to oil sheen compounds
- Support exposure modeling

Quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations

Collaboration

- Support other assessment efforts (see Water, Snow, Ice, and/or Source Oil guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in these guidelines).
- Arctic weather conditions are notoriously variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork; however, realize that it may have to be modified based on actual conditions in the field because sheens are very dynamic. Sampling sheens may be opportunistic if one is encountered during other sampling efforts.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of sites and samples to be collected at each site, taking into account level of effort, potential logistical limitations, weather conditions, and other unanticipated issues that may compromise sample integrity.
- Area-specific modification to this guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of sheen contamination to estimate the number of sampling locations and number of samples per location needed to meet the sampling plan objectives.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant sampling guidelines from the laboratory and consult with them about necessary modifications.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick water-proof labels); solvent rinsing of jars and aluminum foil for polycyclic aromatic hydrocarbons (PAH) analyses, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.

- Sheens are collected for the purpose of determining the origin of the oil, and are useful for: determining the source of the oil when sheens are the only indication of source oil (e.g., diesel spills); fingerprinting sheens of unknown origin (e.g., biogenic, other sources of contamination); and documenting the spatial extent of the spilled oil as it spreads.
- Sheen samples should be collected from specific locations to answer specific questions, such as what is the source of a reported sheen, what is the spatial extent of oiling, or how is the oil on the water surface weathering over time and distance?
- When sampling in remote areas with limited storage and shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised and sample holding times are not exceeded. Remember that it may take multiple days for shipments from rural areas to reach a laboratory facility.
- The number of samples collected need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours, if possible. However, working from a vessel with deck lights that allow safe operations would permit nighttime sampling. This guidance may not apply during winter.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the goals of the sampling plan.

Area selection

- Well-defined study plans and sampling protocols should direct sample collection efforts. If a sampling plan is not available for ephemeral data collection immediately after a spill, sampling should focus on collecting samples of sheens from the spill and other natural or non-spill sources if they are observed.
- Use trajectory models, overflight observations, SCAT observations, or other sources to determine where sheens are likely to be present.
- Opportunistic sheen samples should be collected if sheens are observed during other field data collection activities.
- The number of locations and number of samples per location should be defined in the study design. A minimum guideline for collecting sheen samples is one per sampling location for fingerprinting. Additional samples may be required for other analyses.

Collaboration

- Sheen samples can be collected in conjunction with water/snow, ice, and source oil samples.
- Close collaboration and coordination with other ongoing sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The type of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/collapsible jugs (for storage temperature regulation)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves (preferred) when sampling for sheen under extreme cold conditions
- 125 mL wide-mouth sampling jars – certified organic clean glass jars (solvent rinsed) with Teflon lined lids and labels
- Teflon (PTFE-fluorocarbon polymer) nets/pads (preferred) - 4 inch diameter, and deployment gear (wand, pole, line). Sorbent pads (less ideal)

- Tweezers, hemostats, or pliers; one for each sample, pre-cleaned, wrapped in foil and sealed for transport
- Field Sample Forms, field notebook, chain of custody forms
- Evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Tape measure and ruler
- Packaging materials (bubble wrap and sorbent pads, tape) for glass jars (may be provided by the analytic laboratory)
- Suitable disposal bags for oiled PPE and disposable sampling materials.

Optional (if single-use deployment gear is not available):

- Sufficient quantities of pre-cleaned or disposable deployment gear are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling utensils
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store bought distilled water (less ideal)
  - Laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (account for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.
- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site by leaving the field blank sample jar open for the duration of the sampling period at that site. Record where field blanks were taken on the log sheet.
- Ideally, trip and field blanks are a sampling jar with Teflon sampling material in them.
- If possible, store samples from field/trip blanks in one set of coolers, with oiled samples in a separate set of coolers. If possible, do not include other types of samples in the coolers for the sheen samples; otherwise, take precautions to prevent cross contamination.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will ensure the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be changed between samples to prevent cross contamination or if they become contaminated or damaged. Nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination such as washing gloves with detergent and/or solvents between samples and avoiding contact with the sample material.
- Decontaminate used Teflon net deployment gear (wand/pole, or similar) prior to each use:
  - Wash gear with laboratory-grade detergent and clean water, with a triple clean water rinse (laboratory grade – preferred, distilled water from a local store – less ideal). If distilled water is not available, use “background” water from an up-current clean area
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from deployment gear before use. Do not work with solvents

downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material

- Potential contamination while sampling from vessels (exhaust fumes, oily surfaces) is a very serious concern. Work up-wind of any exhausts, consider sampling sheens from non-motorized craft that is paddled upwind/current from the motorboat. Avoiding sampling from the stern of motorboats.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photos. The numbering sequence of photos uploaded from your camera must not have any gaps (see Field Photography guidelines).
- A description of the characteristics and spatial extent of the sheen that the samples are collected from should be recorded in the field data sheet or the field notebook.
- Sheen samples can be collected from boats, from the shoreline, or by wading in shallow water.
- When sampling through ice:
  - Clear loose ice and snow away from the sampling location and drill through the ice
  - Clean the drill hole area from potential sources of contamination, and allow several minutes for the water to flow freely under the ice before taking a sheen sample
- Teflon nets and pads are the preferred method for sampling sheen as they allow a concentration of light sheens into a relatively small sample size.
- Teflon (PTFE-fluorocarbon polymer) nets (50 to 70 micron-mesh screen, preferable) or pads, can be hand-held, attached to a sampling wand/pole, or attached to the line of a fishing pole. Slowly drag the net or pad through the sheen at least five times or until the net or pads are visibly oiled, and transfer the material into a 125 mL glass container touching the Teflon material as little as possible. To avoid contamination clean tweezers, hemostats, or pliers are to be used to handle the Teflon net or pad. If Teflon nets are not available, the least preferred method would be collection of sheen samples with sorbent pads cut to fit into the sampling jars.
- Minimum guidelines per area are one sample collected with Teflon nets though additional samples may be required.
- Because light PAH fractions are extremely volatile, NEVER split sheen samples after sample collection.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field forms are properly filled out. See appendix A for an example field data form.
- Follow chain of custody procedures for securing samples and complete the Chain of Custody form, noting sample size, sampling device used and any other relevant information for the receiving laboratory in addition to the basic information about the sample indicated on the form. (See Chain of Custody guidelines).
- Record the sample number on both the label and lid. Record the following on the field log sheet:
  - Location of sample collection (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (sheen)
  - Sample #, date/time

  - Sampling method (deployment gear)
  - Note if sample is for QA/QC (field blank, trip blank, rinsate blank)
  - Describe the oiling conditions (using standard shoreline assessment terminology), tidal elevation and weather conditions
  - Characteristics (using standard terminology) and spatial extent of the sheen
  - Characteristics of floating material in the sheen: texture, color, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Maintain strict chain of custody during sample storage and transportation.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately following collection, place all sheen samples in cooler and keep at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Depending on the remoteness of the sampling location, the holding time (7 days) may not be achievable. Under these conditions freeze samples as soon as it is practical to preserve the integrity of the sample.
- Protect the samples from direct sun exposure (e.g., UV radiation).
- Tape lids on sample jars in accordance with chain of custody guidelines and so that they do not accidentally come off.
- Sheen samples should be stored and transported in a separate set of coolers.
- Samples should be stored at 4°C and refrigeration temperature shall be recorded upon sample storage, and monitored and recorded periodically to ensure proper refrigeration.
- Use packing material, such as bubble wrap or sorbent pads, around containers to prevent breakage during handling and shipping. The receiving laboratory may provide packaging materials and shipping containers.
- Ship samples directly to the laboratory as soon as possible with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard ANY samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume and Requirements

Analytical Method	Sample volume	Minimum Detection Levels [a]	Recommended Holding time [b]	Minimum number of samples per location
PAH (including alkylated PAHs) by GC/MS-SIM	Teflon nets or pads	Not applicable	7 days	One Teflon net or pad per location [c]
Petroleum biomarkers (fingerprinting)				

[a] µg/L= ppb; [b] Store at 4 °C (± 1°C) in the dark; [c] Several analyses can be made from a single sample.

Analytical Methods

- **Polynuclear aromatic hydrocarbons** (PAH). Because most of the toxicity in oil is due to PAHs, it is the preferred analysis. It is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard PAH "priority pollutants." This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs, using GC/MS in the selected ion monitoring (SIM) mode. Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. The lab should analyze a sample of the source oil if one is available.
- **Petroleum biomarkers** These compounds are highly resistant to degradation and have a unique distribution for each oil type. Thus, they are valuable for differentiating among different sources of hydrocarbons. However, few laboratories have the capability for quantitative analysis of biomarkers, which is a specialized method using GC/MS in the SIM mode.

Key References

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USEPA. 1986. Test methods for evaluating solid waste. SW 846 Third Edition (and updates).

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Appendix A

Supporting Documentation- Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Oil/Tarball/Water and Snow/Ice sample collection form

Sample Collection Form - OIL/TARBALL/WATER/SNOW/ICE/SHEEN										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling Method**	**Sample Position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(O)il, Tarball (B), (W)ater (SN)ow, (I)ce, (SH)een*	*Sample # (Team ID – sequential number) and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Teflon net, sorbent pad (or other)*	*Collection depth of water sample. Use 0 for surface samples.*	*Volume of sample with units*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD -YYY.YYYYYY*	*Description of sample, equipment used, photo numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										
Samples relinquished by:				**Received by:**						
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**			

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball, Water, Snow, Ice, Sheen	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H) (SN)ow (I)ce (SH)een	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Complete sample IDs are comprised of the following information: Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Guidelines for Collecting Ephemeral Data in the Arctic: WATER

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on the collection of water samples for chemical analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Water samples are generally collected very early in a spill and would rely on vessels of opportunity and emergency go-kits for water sampling offshore. Thus, most sampling to support NRDA will likely be conducted in coastal waters, in conjunction with shore-based assessments, which is what this guideline is focused on.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in the water column
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Study exposure

- Document exposure of water-column organisms to oil compounds
- Support exposure modeling
- Support environmental transport modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations

Collaboration

- Support other ongoing efforts including, but not limited to, validation of remote sensing activities, modeling of exposure and injuries to water-column resources, collection of fluorometry data, and analysis of dissolved vs. particulate oil phases
- Support other assessment efforts (see Intertidal Sediment, Stranded Oil, Ice guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of areas and samples to be collected at each area, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of water contamination or an appropriate power analysis to estimate the number of sampling locations and number of sites per location needed to respond to the sampling objectives.
- For water samples, sampling "areas" can be defined as: 1) waterbodies with defined boundaries (such as lagoons, bays, or river mouths); 2) distances down current from the release site (such as 0-5 km, 5-10 km); and 3) waterbodies that are expected to have similar oil exposure based on observations or models (particularly plume models).
- Depending on the water depth, water samples can be collected at three depths: near surface (0-1 m), mid-depth, and 1 m above the bottom. Generally, near surface samples should be prioritized if the sampling effort is limited by logistics or other factors. In shallower water, samples should be collected at just one near surface depth.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling sites, intertidal zone width, etc. before going into the field. The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with water sampling. If observed during water sampling, tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars for total hydrocarbons (THC) and polycyclic aromatic hydrocarbons (PAH) analyses, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.

- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled and already oiled areas.
- It is important to obtain reliable information and analytical chemistry data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours, if possible. However, working from a vessel with deck lights that allow safe operations would permit nighttime sampling. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.
- Water samples can be collected from boats or by wading in shallow water.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference locations.
- Use trajectory models, conceptual models, overflight information, SCAT data or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be affected by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- Water samples can be collected from marine, estuarine and fresh water habitats. When present, nearshore lagoons with connectivity or potential connectivity to the marine environment should be sampled.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from nearshore water adjacent to locations that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiling water samples from sensitive/productive locations that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Water samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency should be defined in the study design.
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting water samples is at least three samples per waterbody location. If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiling location and two heavily oiled locations.

- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration
- Water samples from nearshore areas can be collected in conjunction with sediment, oil sheen and stranded oil sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field. See Alternative equipment/methods guideline for options if preferred equipment is not available.
- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs− for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for water sampling under extreme cold conditions
- Sampling jars – certified organic-clean glass jars (solvent rinsed) with Teflon-lined lids and labels:
  - 1 L glass jars, amber glass preferred
  - 40 mL septum-capped vials, HCl-preserved preferred, amber glass preferred
- Trip and field blanks – 1 L and 40 mL sampling jars filled with distilled water
- Sorbent pads (for water samples when sheens are present; see Sheen guideline)
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Field notebook, evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Packaging materials (bubble wrap and sorbent pads, tape) for glass jars (may be provided by the analytic laboratory)
- Suitable disposal bags for oiled PPE and disposable sampling materials
- Subsurface water sampler (e.g., Niskin bottle or other) – if needed

Optional (if single-use sampling equipment are not available):
- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (accounts for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.
- Ideally, trip and field blanks are a sampling jar containing ultra-pure or distilled water. Blanks may be provided by the receiving laboratory.
- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site, or at least at an "un-oiled" and "oiled" site, by leaving the field blank sample jar open for the duration of the sampling period at that site. Record the site where field blanks were taken on the field sample form.
- Duplicate samples should be collected at every third sampling site or following the specifications of the work plan. A duplicate sample is collected from the same location and following the same steps as the preceding sample. This is not the same as collecting replicates from each site/depth. Duplicates should account for 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless specified in the work plan.
- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- Water samples for THC and PAH analysis should be placed in certified organic-clean (solvent rinsed) glass containers with Teflon- or aluminum foil-lined lids.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples:
  - To decontaminate the sampler prior to each use, wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. If solvents are not available, use a diluted

detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling materials

- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work upwind of any exhausts, consider sampling from non-motorized craft that is paddled upwind/current from the motorboat, and designate clean areas for sampling. Sampling on the windward side of the vessel is preferred.
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record the sampling site location using a GPS.
- For each sampling site, record:
  - Date, time, weather conditions (e.g., wind direction and speed), and tide level
  - Water depth (in meters) for water samples
  - Presence of biological resources or other relevant information
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- To minimize risks of cross-contamination, collect water samples directly into the sample container by hand (wearing clean disposable Nitrile gloves); a less ideal alternative is to use samplers that can hold 1 L glass bottles. This may be necessary for the collection of subsurface water samples where sampling bottles need to be opened/closed at the targeted water depth.
  - If water samplers (e.g., Niskin bottles) are used these need to be thoroughly decontaminated prior to each use
- Clear surface slicks and sheens prior to deploying the equipment by sweeping the area with a sorbent pad or placing a barrier up-current to divert surface oil around the sampling area, avoiding physical dispersion of the oil into the water column.
- Collect BTEX samples in HCl-preserved 40 mL septum-capped vials. Fill vials completely and cap at the sampling depth or, if using a water sampler, fill the vials to overflow and cap immediately. Vials should not have headspace or air bubbles. If BTEX sampling vials are not available, water samples for THC and PAH should still be collected.
- Collect water samples for THC and PAH in glass containers (organic clean). Leave headspace of about 2 cm for 1 L jars. If sampling directly into jars, fill completely and cap at the sampling depth. Remove the cap only once the sampling jar is no longer in contact with the water and pour out the necessary volume to create headspace before recapping.
- Collect “near surface” water samples at a uniform depth (e.g., 30 cm, which would be up to your elbow if using your hands) below the water surface taking care to avoid any surface slicks or sheens.
- If collecting samples by wading in shallow water, collect samples in waters that are at least 60 cm deep. Collect samples at a uniform depth (e.g., 30 cm, which would be up to your elbow if using your hands) below the surface. Avoid disturbing or suspending bottom material. Stand down current and

wait until any suspended sediment is flushed away before submerging and opening the jar in front of you.

- When sampling by hand:
  - Stand facing the current, if any, and wait until any suspended material is flushed away by the currents
  - Plunge the bottle with the cap on, neck downward, under the water surface in front of you
  - Turn the bottle until the neck points slightly upwards with the mouth directed into the current
  - Uncap the sampling bottle and fill it. Do not touch the cap liner or the inside of the bottle
  - Cap the bottle under water immediately after filling
- When using a sampler:
  - Sampling equipment MUST be deployed and retrieved in the closed position, opening the sampler at the sampling depth
  - All field equipment that comes in contact with the sampling media MUST be thoroughly decontaminated after each sampling event to prevent inadvertent sample contamination
  - If possible, dedicate one set of sampling equipment per degree of oiling to minimize potential cross-contamination
- When sampling through ice:
  - Clear loose ice and snow away from the sampling site and drill through the ice
  - Clean the drill hole area from potential sources of contamination and allow several minutes (~10 min) for the water to flow freely under the ice before taking a sample. If water in the hole freezes over, use clean tools to break through the thin layer of ice and proceed with sample collection (see Ice guideline for additional details)

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each water sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (water)
  - Sample #, date/time
  - Sampling method (directly into jar, sampler)
  - Sample collection depth
  - Note if sample is for QA/QC (field blank, trip blank, rinsate blank)
  - Describe the oiling conditions (using standard shoreline assessment terminology), characteristics of suspended material in the water sample (texture, color, turbidity, biota, vegetation, debris, odor, etc.), distance from shoreline, weather conditions (e.g., wind direction and speed), odors and other relevant information on the field data sheet
- All sample numbers must be unique. Use the sample number convention provided by data management if one is available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution on the water surface is best accomplished with photography, video, and good field notes. Samples may be needed for fingerprinting or monitoring weathering, to correlate a

degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes (see relevant guidelines for details about sampling oil).

- Document presence of slicks, weather, wave conditions, etc. which might suggest mixing of surface oil during sampling.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately place all water samples in a cooler and keep at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature if heated storage space is not available. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Protect the samples from direct sun exposure (e.g., UV radiation).
- Tape lids on sample bottles so that they do not accidentally come off.
- If possible, store samples from unoiled areas in one set of coolers, with oiled samples in a separate set of coolers.
- THC and PAH: can add 1 mL of 6 N HCl/liter of sample within 2 hours of sampling to inhibit microbiological activity. Not required by EPA.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping. The receiving laboratory may provide packaging materials and shipping containers.
- Water samples can be held at 4°C in the dark for up to 7 days (includes recommended holding time in the field and receiving laboratory) without loss of sample integrity. Samples should not be frozen.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels [a]	Recommended Holding Time [b]	Minimum No. of Samples per Location
BTEX [c] (full scan mode)	40 mL	10 µg/L	7 days; 14 days with preservatives	1 per depth
BTEX (SIM)	1 liter [d]	0.1-1 µg/L	7 days	1 per depth
Total Hydrocarbons (THC) by GC/FID		15 µg/L		
PAH (including alkylated PAHs) by GC/MS-SIM		0.001-0.01 µg/L		
Chemical biomarkers (fingerprinting)		0.001-0.01 µg/L		

[a] µg/L= ppb; [b] Store at 4°C in the dark; [c] Sometimes referred to as Volatile Aromatic Hydrocarbons (VAH) or Volatile Organic Compounds (VOC); BTEX are a subset of VAHs/VOCs; [d] Several analyses can be made from a single sample.

Analytical Methods

- **Polynuclear aromatic hydrocarbons** (PAH). Because most of the toxicity in oil is due to PAHs, it is the preferred analysis. It is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard 16 PAH "priority pollutants." This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated, using GC/MS in the selected ion monitoring (SIM) mode. Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. The lab should analyze a sample of the source oil as well.
- **Chemical biomarkers**. These chemicals are the most important hydrocarbon groups used for chemical fingerprinting allowing a quantitative identification of the source oil. Because biomarkers are more resistant to weathering and biodegradation than other hydrocarbons, these can also be used to quantify the degree of oil weathering. Chemical biomarkers are typically analyzed by GC-FID (e.g., EPA Method 8015 Modified). These chemicals are typically analyzed concurrently with THCs.
- **Volatile organic hydrocarbons** (benzene, toluene, ethylbenzene, and xylene, or BTEX). For oil spill applications, the standard EPA Method 8240/8260 (purge & trap) should be modified by running the GC/MS in selected ion monitoring or full scan mode to include the higher alkylated (C3 and C4) benzenes. Detection limits should be 1 ppb for individual analytes; 0.1 ppb is easily achievable in SIM mode.
- **Total hydrocarbons** (THC). Often referred to as total petroleum hydrocarbons (TPH), but most methods do not differentiate among petroleum, petrogenic, and biogenic hydrocarbons. THC by GC-FID (total area of FID gas chromatogram of combined f_1 and f_2 fractions after column chromatography; e.g., EPA Method 8015 Modified) is often the preferred method because of the low detection limit (compared to other THC methods) and the direct measurement of hydrocarbons. This method does not detect low boiling point compounds (below n-C_8). THC analyses generally will not provide the data needed to support calculation of toxic effects from PAH exposure, and will have to be corrected to equivalent PAHs. The THC results, however, can be used to document changes in oil weathering and map extent of exposure of water column resources, if meaningful detection limits are used (15 µg/L). THC results can be used as a screening tool to estimate the presence and amount of hydrocarbons and provide an indication of which samples should receive highest priority for more extensive analyses.

Key References

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Appendix A
Supporting Documentation- Field Data Forms

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form:

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Oil/Tarball/Water/Snow/Ice/Sheen Sample Collection Form

Sample Collection Form - OIL/TARBALL/WATER/SNOW/ICE/SHEEN										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling Method**	**Sample Position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(O)il, Tarball (B), (W)ater or (SN)ow, (I)ce, (SH)een*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Method of sampling (i.e., sampler or other)*	*Collection depth of water sample. Use 0 for surface samples*	*Volume of sample with units*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD -YYY.YYYYYY*	*Description of sample, equipment used, photo numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										
Samples relinquished by:				**Received by:**						
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**		**Print Name - Sample Runner/ Command Post**		

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball, Water, Snow, Ice, Sheen	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H) (SN)ow (I)ce (SH)een	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Complete sample IDs are comprised of the following information: Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Guidelines for Collecting Ephemeral Data in the Arctic: SNOW

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on the collection of snow from shoreline habitats for chemical analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. This guideline can be used if snow-covered shorelines are impacted by oil or if snow is deposited on top of an oiled shoreline.

Sampling Objectives

Characterize oil

- Determine the amount and composition of oil compounds in snow on shorelines
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and amount of oiling in shoreline habitats
- Document the transport and fate of oiled snow in shoreline and onshore habitats

Study exposure

- Document exposure of shoreline and terrestrial organisms to oil
- Support exposure modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations
- Support environmental transport modeling

Collaboration

- Support other ongoing efforts including, but not limited to, validation of remote sensing activities, modeling of exposure and injuries to terrestrial and shoreline resources, assessment of oil transport and fate
- Support other assessment efforts (see Intertidal Sediment and Stranded Oil guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of shoreline contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per location needed to respond to the sampling objectives. If wind transport of oil and oiled snow is a possibility, use an appropriate model to estimate transport distance and direction.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling sites, shoreline characteristics, etc. before going into the field. This is particularly important when collecting snow from shoreline areas.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with snow sampling. If observed during snow sampling, water, tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars for total hydrocarbons (THC) and polycyclic aromatic hydrocarbons (PAH) analyses, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.

- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable information and analytical chemistry data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- This guideline is only relevant to habitats and areas where snow is present.
- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- Consider that oil may not be visible if snow has been deposited on top of it. Additionally, oil and oiled snow may be transported away from shorelines by wind so the oiled area could be much larger than the shoreline and extent and amount of oiling may be very dynamic.
- Samples should also be collected from locations known or suspected to be affected by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from shorelines that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting snow from sensitive/productive sites that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Snow samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency should be defined in the study design.
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting snow samples per location of relatively uniform oiling exposure. If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiling location and two heavily oiled locations.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area. Consider possible transport of oil and oiled snow by wind; this could cause exposure gradients that extend into the onshore area.

Collaboration

- Snow samples can be collected in conjunction with sediment, water and stranded oil sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field. See Alternative equipment/methods guideline for options if preferred equipment is not available.

- Coolers – for sample storage and transport
- Ice packs– for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Teflon (PTFE) bags – 60 x 60 cm, organic clean (solvent rinsed) with closures or cable ties (preferred) or metal sampling cans with lids – 4 L minimum volume, organic clean (less ideal)
- Shovel
- Trip and field blanks – 1 L and 40 mL sampling jars filled with distilled water
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Field notebook, evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Tape measure and ruler
- Packaging materials for glass jars – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment are not available):

- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (accounts for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.
- Ideally, trip and field blanks are a sampling jar containing ultra-pure or distilled water. Blanks may be provided by the receiving laboratory.
- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site, or at least at an "un-oiled" and "oiled" site, by leaving the field blank sample jar open for the duration of the sampling period at that site. Record the site where field blanks were taken on the field sample form.
- Duplicate samples should be collected at every third sampling site or following the specifications of the work plan. A duplicate sample is collected from the same location and following the same steps as

the preceding sample. This is not the same as collecting replicates from each site. Duplicates should account for 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless specified in the work plan.

- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- Snow samples for THC and PAH analysis should be placed in organic-clean (solvent rinsed) Teflon (PTFE) bags or organic clean metal sampling cans with water-tight closures.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples:
  - To decontaminate the shovel prior to each use, wash with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Allow solvents to evaporate from shovel before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling materials
- Potential sources of contamination while sampling (exhaust fumes, oily surfaces, cigarette smoke) are a concern. Exhaust can leave remnant contaminants in snow even after the engine has been shut off and the smell has dissipated. Consider wind direction when transiting or approaching sampling sites to avoid permanently contaminating a site, work upwind of any exhausts, and designate clean areas for sampling. Avoid collecting samples near snow machine, ATV, or other motorized vehicle tracks unless intentionally documenting other sources of contamination.
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Study Design Implementation

- Record the sampling site using a GPS.

- For each sampling site, record:
  - Date, time, air temperature, weather conditions (e.g., wind direction and speed), and tide level
  - Snow depth and characteristics
  - Extent and degree of visible oiling (use SCAT guidelines for a more detailed assessment of shoreline oiling if needed)
  - Presence of ice along shoreline
  - Presence of biological resources or other relevant information

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline). Use a photo scale or other object when taking pictures of snow.
- If snow depth is less than 30 cm, measure and record the depth. Use a clean shovel to collect a sample of snow that is approximately cubic in shape and includes the entire depth of the snow pack. Avoid sampling the sediments underlying the snow. The dimensions of the sample pit will depend on the depth of the snow pack; more area will be sampled for a shallower snow pack to achieve the desired sample volume. Fill the Teflon bag approximately ¾ full, twist the bag shut and secure with a closure or at least two cable ties. Do not overfill the sample bag. It may be necessary to have one person shovel and one person hold the bag. If using metal cans, fill the cans with at least 4 L of snow and close securely.
- If the snow depth is greater than 30 cm, use a clean shovel to dig a snow pit that exposes a vertical slice of the snow pack from the snow surface to the ground. If possible, avoid exposure of the sampling wall to direct sunlight. Take picture of the pit and record any visible oiling. Samples can be collected at discreet depths, starting from the surface of the snow pack. Record the depth and fill a sample bag or sample can as described above. Alternately, a composite sample can be collected by cutting a narrow column of snow from the top of the snow pack to just above ground along the snow pit face with the shovel. This can be repeated as many times as necessary to fill the sample bag or can. Seal the sampling container as described above.
- Sealed sample bags should be protected from sunlight and maintained frozen or at less than 4°C. If ambient temperature is less than 4°C, no refrigeration is necessary in the field.
- Make sure to clean the shovel completely between each sample or use a disposable sampling scoop.
- When sampling heavily oiled snow, follow the Source Oil guidelines to collect oil samples.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each snow sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.

- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample label and lid/closure. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (snow)
  - Sample #, date/time
  - Sampling method (directly into container)
  - Sample collection depth
  - Note if sample is for QA/QC (field blank, trip blank, rinsate blank)
  - Describe the oiling conditions (using standard shoreline assessment terminology), distance from shoreline, weather conditions (e.g., wind direction and speed), odors and other relevant information on the field data sheet
- All sample numbers must be unique. Use the sample number convention provided by data management if one is available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution in snow is best accomplished with photography, video, and good field notes. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Document presence of slicks, weather, wave conditions, etc. which might suggest mixing of surface oil during sampling.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- When prioritizing ephemeral data sample collection, take into account the recommended holding time for snow samples and plan accordingly, depending on the feasibility of samples being received for analysis within the 7-day thawed holding time.
- If possible, maintain snow samples frozen. If freezer space is unavailable for storage and/or frozen transport is not an option, maintain samples at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Protect the samples from direct sun exposure (e.g., UV radiation).
- Tape lids on sample cans so that they do not accidentally come off.
- If possible, store samples from unoiled areas in one set of coolers, with oiled samples in a separate set of coolers.
- Frozen snow samples can be held for up to 30 days. Melted snow samples can be held at 4°C in the dark for up to 7 days (includes recommended holding time in the field and receiving laboratory) without loss of sample integrity.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage or puncture, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels [a]	Recommended Holding Time [b]	Minimum No. of Samples per Location
Total Hydrocarbons (THC) by GC/FID	1 liter [d]	15 µg/L	30 days (frozen) 7 days (at 4°C)	3
PAH (including alkylated PAHs) by GC/MS-SIM		0.001-0.01 µg/L		
Chemical biomarkers (fingerprinting)		0.001-0.01 µg/L		

[a] µg/L= ppb; [b] Store in the dark; [c] Sometimes referred to as Volatile Aromatic Hydrocarbons (VAH) or Volatile Organic Compounds (VOC); BTEX are a subset of VAHs/VOCs; [d] Liquid volume. Several analyses can be made from a single sample.

Analytical Methods

- **Polynuclear aromatic hydrocarbons** (PAH). Because most of the toxicity in oil is due to PAHs, it is the preferred analysis. It is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard 16 PAH "priority pollutants." This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs (approximately 43 PAHs), using GC/MS in the selected ion monitoring (SIM) mode. Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. The lab should analyze a sample of the source oil as well.
- **Chemical biomarkers**. These chemicals are the most important hydrocarbon groups used for chemical fingerprinting allowing a quantitative identification of the source oil. Because biomarkers are more resistant to weathering and biodegradation than other hydrocarbons, these can also be used to quantify the degree of oil weathering. Chemical biomarkers are typically analyzed by GC-FID (e.g., EPA Method 8015 Modified). These chemicals are typically analyzed concurrently with THCs.
- **Total hydrocarbons** (THC). Often referred to as total petroleum hydrocarbons (TPH), but most methods do not differentiate among petroleum, petrogenic, and biogenic hydrocarbons. THC by GC-FID (total area of FID gas chromatogram of combined f_1 and f_2 fractions after column chromatography; e.g., EPA Method 8015 Modified) is often the preferred method because of the low detection limit (compared to other THC methods) and the direct measurement of hydrocarbons. This method does not detect low boiling point compounds (below n-C_8). THC analyses generally will not provide the data needed to support calculation of toxic effects from PAH exposure will have to be corrected to equivalent PAHs. The THC results, however, can be used to document changes in oil weathering, and map extent of exposure of water column resources, if meaningful detection limits are used (15 µg/L). THC results can be used as a screening tool to estimate the presence and amount of hydrocarbons and provide an indication of which samples should receive highest priority for more extensive analyses.

Key References

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Appendix A
Supporting Documentation- Field Data Forms

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form:

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Oil/Tarball/Water/Snow/Ice/Sheen Sample Collection Form

Sample Collection Form - OIL/TARBALL/WATER/SNOW/ICE/SHEEN										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling Method**	**Sample Position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(O)il, Tarball (B), (W)ater or (SN)ow, (I)ce, (SH)een*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Method of sampling (i.e., sampler or other)*	*Collection depth of water sample. Use 0 for surface samples*	*Volume of sample with units*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD -YYY.YYYYYY*	*Description of sample, equipment used, photo numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										

Samples relinquished by:				**Received by:**			
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball, Water, Snow, Ice, Sheen	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H) (SN)ow (I)ce (SH)een	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Complete sample IDs are comprised of the following information: Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Guidelines for Collecting Ephemeral Data in the Arctic: INTERTIDAL SEDIMENTS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collection of intertidal sediment samples for chemical analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. See other guidelines for collection of biological samples from different types of intertidal habitats.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in intertidal sediments compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and degree of intertidal sediment oiling
- Measure sediment characteristics for interpreting chemical and biological results
- Support oil environmental transport modeling by documenting where oil stranded onshore

Study exposure

- Document exposure of sediment dwelling organisms to oil compounds
- Support exposure modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations

Collaboration

- Support other assessment efforts (see Subtidal Sediment, Stranded Oil, Water, Snow, Ice, Shellfish Tissue, and/or Sand Beach Infauna guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Intertidal sediments are difficult to sample because of the inherent heterogeneity of oil distribution over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as intertidal habitats or lagoons
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of intertidal sediment contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per site needed to respond to the sampling objectives.
- A stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected, is recommended if no other sampling strategy has been developed. This type of sampling improves the representative quality of samples by reducing sampling error (variability).
- Unreplicated grab samples are not very useful to injury quantification. Samples should be quantitative relative to a surface area metric.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling transects, transect spacing, intertidal zone width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with intertidal sediment sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars

and aluminum foil for total hydrocarbons (THC) and polycyclic aromatic hydrocarbons (PAH) analyses, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable information and analytical chemistry data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Areas for intertidal sediment sampling are coastal areas with relatively fine sediments (e.g., clay, silt, sand, granules, or small pebbles). Sediments from inundated tundra and peat shorelines can also be sampled though some modifications to the guidelines may be necessary. Quantification metrics may differ and the receiving analytical laboratory will need to be informed of the nature of the sample. See the Gravel Beach and Rocky Beach guidelines for information about sampling intertidal habitats with larger sediment sizes.
- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data or other tools to determine what location have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled intertidal areas that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiled intertidal sediments from sensitive/productive areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.

- Sediment samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency is a function of oil persistence, biological community, habitat importance, and resource availability and should be defined in the study design.
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting sediment samples is at least three samples per intertidal zone per location of relatively uniform oiling exposure. If relevant data are available, a power analysis or other modeling approaches should be used to determine the number of samples needed before going into the field.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration

- Sediment samples can be collected in conjunction with nearshore water sampling, stranded oil sampling, and sand beach infauna surveys.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/collapsible jugs – for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for sediment sampling under extreme cold conditions
- Sampling jars – 4 or 8 oz certified organic-clean jars with Teflon-lined lids and labels – for TPH/PAH/biomarkers; for fine sediments
- Aluminum foil: the dull side should be pre-cleaned with acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) to wrap samples of larger gravel, with the dull side in contact with the sample
- 10 mL soap-cleaned glass or plastic containers – for total organic carbon, TOC
- Ziploc or Whirl-Pak bags – for grain size
- Ziploc bags and additional sampling jars – for non-viscous tarballs or oil residues
- Shovel
- Stainless steel spatulas and spoons – organic clean, wrapped in foil and sealed for transport
- Disposable aluminum pans – for composite samples
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Field notebook, evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Tape measure and ruler
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment is not available):

- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (accounts for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.
- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site, or at least at an "unoiled" and "oiled" site, by leaving the field blank sample jar open for the duration of the sampling period at that site. Record the site where field blanks were taken on the field sample form.
- Ideally, kiln-fired sand supplied by the laboratory can be transferred (poured or scooped) from one jar to another and returned to the lab as a field blank, but if this is not possible, use the open-jar technique (empty jar).
- Duplicate samples should be collected at every third sampling site or following the specifications of the work plan. A duplicate sample is collected from the same location and following the same steps as the preceding sample. This is not the same as collecting replicates from each site/depth. Duplicates should account for 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless specified in the work plan.
- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).

- The only equipment to be used between sites is a shovel, which should be cleaned with soap and clean water. Repeated digging in clean sediments can be a last resort for cleaning the shovel if soap or clean water are not available. Alternatively, use a clean dry towel or other dry material to clean the shovel before its next use. Additional cleaning may be required when working at oiled sites (see below).
- Sediment samples for THC and PAH analysis should be placed in certified organic-clean (solvent rinsed) glass containers with Teflon- or aluminum foil-lined lids. Samples for TOC can be placed in soap-cleaned glass or plastic containers. For grain size, Ziploc or Whirl-Pak bags can be used.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.
- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.

Study Design Implementation

- If no other sampling strategy has been developed, use a stratified random sampling approach by randomly selecting the first transect starting point and expanding the sampling site (other transects) systematically into a grid from that initial point. Randomly select sampling sites within each intertidal zone intervals.
- Based on the study design and/or sampling strategy outlined before going into the field, establish a minimum of three transects spaced at least 30 m apart (recommended) (Figure 1). Transects should be perpendicular to the shoreline and encompass the entire intertidal zone. Run transects within two hours of low tide (before or after). In the Chukchi and Beaufort seas the tide range is small (<30 cm); wind-driven storm surge is the most determinant factor in water height and should be considered for sampling. Tides are only a consideration south of the Bering Strait where the tidal range is greater than 1 m.
  - Record the transect location using a GPS and accurately plot the transect location on a map or aerial photograph
  - If possible, permanently mark the transect location using "front" and "back" stakes that line up with the transect. Consider placement carefully to minimize loss due to vandalism, erosion, ice scouring, etc.
  - Record the transect angle with a compass so it can be re-surveyed at a later date even if one of the stakes is lost; note whether the angle is magnetic or true north

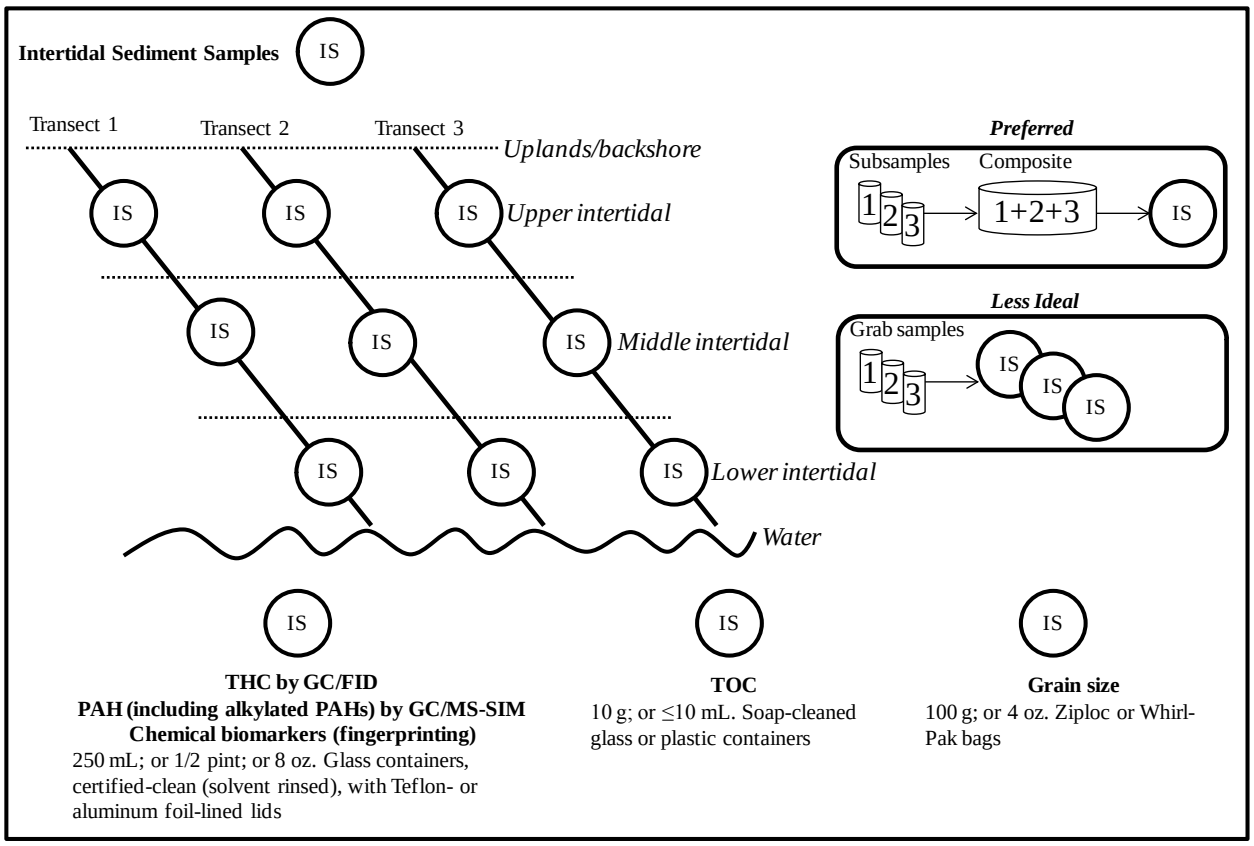

Figure 1. Schematic representation of the recommended intertidal sediment sampling strategy, including transects and sampling sites. Dashed lines represent approximate tidal zones. IS= intertidal sample. Area-specific modifications may be needed. For example at sites with very narrow intertidal zones, only one or two sampling intervals may be used.

- Divide each transects into sampling intervals based on the intertidal zones: upper intertidal, middle intertidal and lower intertidal.
- On some high-Arctic shorelines, the intertidal zone may be very narrow. If an intertidal transect is too narrow to have three sampling intervals on, consider running the transect from the supratidal or storm surge line (usually demarcated by a line of logs or debris) to the lower intertidal. Alternately, two sampling intervals can be defined on each transect, in the upper (or storm surge) and lower intertidal zone, or just one station if the intertidal zone is very narrow.
- For each transect, record:
  - Date, time, weather conditions (e.g., wind direction and speed), and tide level
  - Physical setting (shoreline orientation, exposure, etc.)
  - Length of the transect (in meters) and of the sampling zone
  - Sediment type
  - Extent and degree of visible shoreline oiling (use SCAT guidelines for a more detailed assessment of shoreline oiling if needed)
  - Extent and degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Presence of snow or ice along shoreline
  - Presence of biological resources or other relevant information
- Collect samples from each intertidal zone (described below) and record the distance along the transect and GPS coordinates of each sampling site.

- If specified in the work plan, collect separate splits of homogenized samples for infauna or toxicity testing, so they can be correlated with chemical results.
- Take pictures of the transect before and after sampling and pictures of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If intertidal sediments are covered by snow or ice, carefully remove the snow/ice without mixing or disturbing the sediment underneath and proceed with sediment sampling. Note snow and ice conditions on the field data sheet. If snow or ice in the intertidal zone are impacted by oil (as opposed to deposited on top of oiled sediments), it may be desirable to collect snow/ice samples in addition to sediments for chemical analysis (see the Ice guideline).
- Special consideration should be given to sampling frozen sediments. Sediments frozen *in situ* (<-7°C) must be maintained frozen in appropriate containers and sub-sampled as soon as practical without thawing. Consider using a sturdy shovel, rock pick, or other tool to loosen sediments for sampling. Collection of frozen samples should be noted in the field logbook.
- Carefully remove gravel, sticks, and other debris on the sediment without mixing or disturbing the sediment underneath. For surface sediments, use a clean spatula to accurately collect the top 2 cm.
- Composite samples (of at least 3 subsamples) are preferred for characterization of a sampling site. Subsamples should be collected at random within a 5 m radius and within the same tidal zone or sampling interval (see Figure 1). Mix and homogenize composite samples before placing in the jar(s) using a disposable aluminum pan. Collect a minimum of 3 replicates per site. A less preferable alternative, if it is not feasible to composite samples in the field, is to collect a minimum of 3 grab samples at each sample site (interval on a transect).
- To collect surface samples from inundated tundra, use a metal spatula or shovel to cut the vegetative mat and collect a sample of the top layer of vegetation and sediments. Consider surveying the extent and degree of oiling and/or collecting stranded oil samples (see Stranded Oil guideline) if it is not possible to collect quantitative inundated tundra intertidal sediment surface samples. To collect subsurface samples in coarse sediments (sand and gravel), it is easiest to use a shovel to dig a small trench/pit and collect the desired sediment intervals from the exposed wall in the trench/pit.

- To collect subsurface samples in fine sediments (e.g., clay, silt, mud) use a shovel to dig out a slice of sediments to the desired depth. Collect the sample of the entire oiled layer from the natural break side of the shovel-full of sediments (see photograph). For example, if the oil penetrated from the surface to 10 cm, collect a representative sample of this entire layer; if the oiled layer was at a depth of 5-10 cm, collect a representative sample of this entire layer.

- Though oil is unlikely to penetrate inundated tundra shorelines, subsurface sediments samples can be collected in tundra by digging a small pit and collecting the desired sediment intervals from the exposed wall in the pit.
- For subsurface samples, record the depth of each sediment interval, total depth of the pit, details about the vertical profile including distinct layers, type of material and presence of biological structures, degree of sample disturbance, and the presence of oil, oily sheen and/or the smell of oil.
- In addition to collecting samples for chemical analysis, take samples for TOC (placed in soap-cleaned glass or plastic containers) and grain size (placed in Ziploc or Whirl-Pak bags) analysis.
- Collect representative non-viscous tarball or oil residues samples, which can be scooped into sampling jars using a stainless steel spatula or spoon, or wrapped in aluminum foil and double bagged in Ziploc bags with sample ID label placed between the inner and outer bags.
- Photograph the sampling pit (see Field Photography guideline).
- Clean the spade of the shovel with soap and water between each sampling pit to prevent cross contamination.
- Discrete samples from a single sample point may be collected to represent a specific condition, such as a tarball for fingerprinting and source identification (see Stranded Oil guideline).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each intertidal sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (sediment, soil, tundra)
  - Sample #, date/time
  - Sampling method (grab, core, composite) and sample collection depth
  - Note if sample is for QA/QC (field blank, trip blank, rinsate blank) or if it is a spilt or duplicate sample
  - Sediment oiling conditions (using standard shoreline assessment terminology), tidal elevation, weather conditions (e.g., wind direction and speed), sediment characteristics, vertical changes in sediment characteristics, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.; vertical changes in sediment characteristics
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- If sample volume is split between two jars, both jars should receive the same sample ID and be recorded on a single line of the Chain of Custody form.
- Documenting oil distribution in intertidal areas is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.

- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately place all sediment samples for chemical analysis in a cooler and keep at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature if heated storage space is not available. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Refrigerate (do not freeze) samples for TOC. Samples for grain size do not require refrigeration.
- Protect samples for chemical analysis from direct sun exposure (e.g., UV radiation).
- Tape lids on sample bottles so that they do not accidentally come off.
- If possible, store samples for chemical analysis from unoiled locations in one set of coolers, with oiled samples in a separate set of coolers.
- Samples for TOC and grain size can be stored separately from samples for chemical analysis.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping. Take special care with gravel sediments because individual pieces can rattle around during shipping and break the glass jars.
- Freeze samples for chemical analysis as soon as practical or by the end of each day if samples are not going to be analyzed within 7 days of collection.
- If sampling sediments frozen in situ (<-7°C), samples MUST be maintained frozen in appropriate containers and sub-sampled as soon as practical without thawing.
- Sediment samples can be held frozen in the dark for several years without loss of sample integrity.
- Sediment extracts can be held at 4°C in the dark for 40 days without loss of sample integrity.
- Ship samples directly to the lab as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed, and use ice packs or dry ice to maintain storage temperatures during shipment.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels [a]	Recommended Holding Time	Minimum No. of Samples per Location
Total Hydrocarbons (THC) by GC/FID	250 mL; or 1/2 pint; or 8 oz. Filled ¾ full [c]	15 µg/kg	7 days at 4°C [b] or several years frozen[c]	1 per site
PAH (including alkylated PAHs) by GC/MS-SIM		0.001-0.01 µg/kg		
Petroleum biomarkers (fingerprinting)		0.001-0.01 µg/kg		
Total Organic Carbon (TOC)	10 g; or ≤10 mL Filled ¾ full	0.01%	28 days	1 per site
Grain size	100 g; or 4 oz	NA	NA	1 per site

[a] µg/L= ppb; [b] Store at 4°C in the dark; [c] Stored frozen in the dark; [d] Several analyses can be made from a single sample.

Analytical Methods

- **Total hydrocarbons** (THC). Often referred to as total petroleum hydrocarbons (TPH), but most methods do not differentiate among petroleum, petrogenic, and biogenic hydrocarbons. THC by GC-FID (total area of FID gas chromatogram of combined f_1 and f_2 fractions after column chromatography; e.g., EPA Method 8015 Modified) is often the preferred method because of the low detection limit (compared to other THC methods) and the direct measurement of hydrocarbons. This method does not detect low boiling point compounds (below n-C_8). THC analyses generally will not provide the data needed to support calculation of toxic effects from PAH exposure, and will have to be corrected to equivalent PAHs. The THC results, however, can be used to document changes in oil weathering and map extent of exposure of water column resources, if meaningful detection limits are used. THC results can be used as a screening tool to estimate the presence and amount of hydrocarbons and provide an indication of which samples should receive highest priority for more extensive analyses.
- **Polycyclic aromatic hydrocarbons** (PAH). Because most of the toxicity in oil is due to the PAHs, it is often the preferred analysis. However, PAHs are expensive and require special laboratory skills. If PAHs are to be measured, it is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard PAH "priority pollutants". This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs, using GC/MS in the selected ion monitoring (SIM) mode. Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. Have the lab also run the source oil with each batch.
- **Petroleum biomarkers**. These chemicals are the most important hydrocarbon groups used for chemical fingerprinting allowing a quantitative identification of the source oil. Because biomarkers are more resistant to weathering and biodegradation than other hydrocarbons, these can also be used to quantify the degree of oil weathering. Biomarkers include steranes/triterpanes, which are "fossil" compounds unique to the oil formation and compounds that provide a secondary and confirming line of evidence in forensic oil identification. Chemical biomarkers are typically analyzed by GC-FID (e.g., EPA Method 8015 Modified). These chemicals are typically analyzed concurrently with THCs.
- Analyses may also include:
  - **Sediment grain size**. There are different methods for measuring sediment grain size and may include sieve analysis of gravel to sand fractions, pipette analysis for muddy sediments, or more sophisticated methods that use rapid sediment analyzer equipment
  - **Water content**. Water content is typically measured in conjunction with chemical and TOC analyses by drying a specified mass of sediment in an oven at constant temperature (typically 100-110 °C) and calculating the mass difference between wet and dry sediments
  - **Total organic carbon**. There are different methods for measuring TOC in sediments, and may include high-temperature combustion methods or more sophisticated methods that use total organic carbon analyzers

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Appendix A
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and initialing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Soil/Sediment Sample Collection Form

Sample Collection Form - SOIL/SEDIMENT										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number**	**Sample Time**	**Sampling Method**	**Sample Collection Depth**	**Depth units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(S)ediment, Soil (L), Tundra (T)*	*Sample # (Team ID – sequential number)*	*(24-hr clock, local time)*	*Method of sampling (i.e., core, grab or composite)*	*Include upper and lower collection depth. Use 0 for surface samples.*	*Units for depth values*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD-YYY.YYYYYY*	*Description of sample, equipment used, including estimated volume, photo numbers, etc.*

Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)

Samples Relinquished by:				**Received by:**			
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment	(GR)ab	(c)m	
Soil (L)	(CO)re	(m)	
(T)undra	Co(MP)osite	(i)nches	
Blan(K) Water		(f)eet	
Oil, Tarball or Water	**Sampling Method**	**Sample Position/Depth**	
(O)il	(GR)ab	(FLOAT)ing	
Tarball (B)	(SC)rape	(SUB)merged	
(W)ater	(OT)her	(STRAND)ed	
Blan(K) Water		(COV)ering	
Other (H)		0 - (Surf)ace	
		<depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue	(WH)ole body	(MU)scle	
Wrack (R)	Whole body w/o shell (WNS)	Yolk	
Blan(K) Water	Chorioallantoic Membrane (CAM)	NA <for Wrack only>	
	Egg (EM)bryo		
	Fillet with skin (FS)	**Species**	
	Fillet without skin (FWOS)	<enter species>	
	Gall Bladder (GB)	NA <for Wrack only>	
	Leaves (LEV)		
	Leaves and stems (LVS)		
	(LI)ver		
Sample Identifier system			
Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			
QA/QC types:		Other sample types:	
Field Blank (FB)	Rinsate Blank (RB)	(S)plit	
Trip Blank (TB)	(D)uplicate		

Guidelines for Collecting Ephemeral Data in the Arctic: SUBTIDAL SEDIMENTS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on the collection of subtidal sediment samples for chemical analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in subtidal sediments compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and degree of subtidal sediment oiling
- Measure sediment characteristics for interpreting chemical and biological results
- Support oil environmental transport modeling

Study exposure

- Document exposure of sediment dwelling organisms to oil compounds
- Support exposure modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations

Collaboration

- See guidelines for biota and aquatic vegetation for information about documenting and collecting biological data in subtidal sediments
- Support other assessment efforts (see Intertidal Sediment and Water guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Subtidal sediments are difficult to sample for oil contamination because of the inherent heterogeneity of oil distribution over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of subtidal sediment contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per site needed to respond to the sampling objectives.
- Unreplicated grab samples are not very useful to injury quantification. Samples should be quantitative relative to a surface area metric.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with subtidal sediment sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars and aluminum foil, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding shipment of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.

- It is important to obtain reliable information and analytical chemistry data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours, if possible. However, working from a vessel with deck lights that allow safe operations would permit nighttime sampling. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference locations.
- When present, sediments from nearshore lagoons with connectivity or potential connectivity to the marine environment should be sampled.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- Use charts, ShoreZone, and other data about benthic habitat and seafloor substrate (data layers available in Arctic ERMA) to select oiled and reference sampling areas.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled subtidal locations that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiled subtidal sediments from sensitive/productive locations that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled "control" locations and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Sediment samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency is a function of oil persistence, biological community, habitat importance, and resource availability and should be defined in the study design.
- The number of locations and number of sites per location should be defined in the study design. A <u>minimum</u> guideline for collecting sediment samples is at least three subtidal sites per location of relatively uniform oiling exposure. If relevant data are available, a power analysis or other modeling approaches should be used to determine the number of samples needed before going into the field.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration

- Sediment samples can be collected in conjunction with nearshore water sampling.
- Subtidal sediments are generally collected from a boat. In the spring, samples can be collected through the landfast ice.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for sediment sampling under extreme cold conditions
- Sampling jars – 4 or 8 oz certified organic-clean jars with Teflon-lined lids and labels − for TPH/PAH/biomarkers; for fine-grained sediments
- 10 mL soap-cleaned glass or plastic containers − for TOC
- Ziploc or Whirl-Pak bags − for grain size samples
- Appropriate sampling devices. Examples include: modified van Veen grab, Ekman grab, and box dredge
- Coring tubes – single-use, disposable preferred
- Flat scoops (stainless steel or plastic)
- Stainless steel spatulas and spoons – organic clean, wrapped in foil and sealed for transport
- Disposable aluminum pans − for composite samples
- Aluminum foil
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Field notebook, evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment is not available):

- Sufficient quantities of pre-cleaned or disposable single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (accounts for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.

- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site, or at least at an "unoiled" and "oiled" site, by leaving the field blank sample jar open for the duration of the sampling period at that site. Record the site where field blanks were taken on the field sample form.
- Ideally, kiln-fired sand supplied by the laboratory can be transferred (poured or scooped) from one jar to another and returned to the lab as a field blank, but if this is not possible, use the open-jar technique (empty jar).
- Duplicate samples should be collected at every third sampling site or following the specifications of the work plan. A duplicate sample is collected from the same location and following the same steps as the preceding sample. This is not the same as collecting replicates from each site/depth. Duplicates should account for 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless specified in the work plan.
- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- Sediment samples for THC and PAH analysis should be placed in certified organic-clean (solvent rinsed) glass containers with Teflon- or aluminum foil-lined lids. Samples for TOC can be placed in soap-cleaned glass or plastic containers. For grain size, Ziploc or Whirl-Pak bags can be used.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples, including dredges:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-

contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.

Study Design Implementation

- Based on the study design and/or sampling strategy outlined before going into the field, collect samples from at least 3 subtidal sites in areas of relatively uniform oiling conditions (Figure 1).

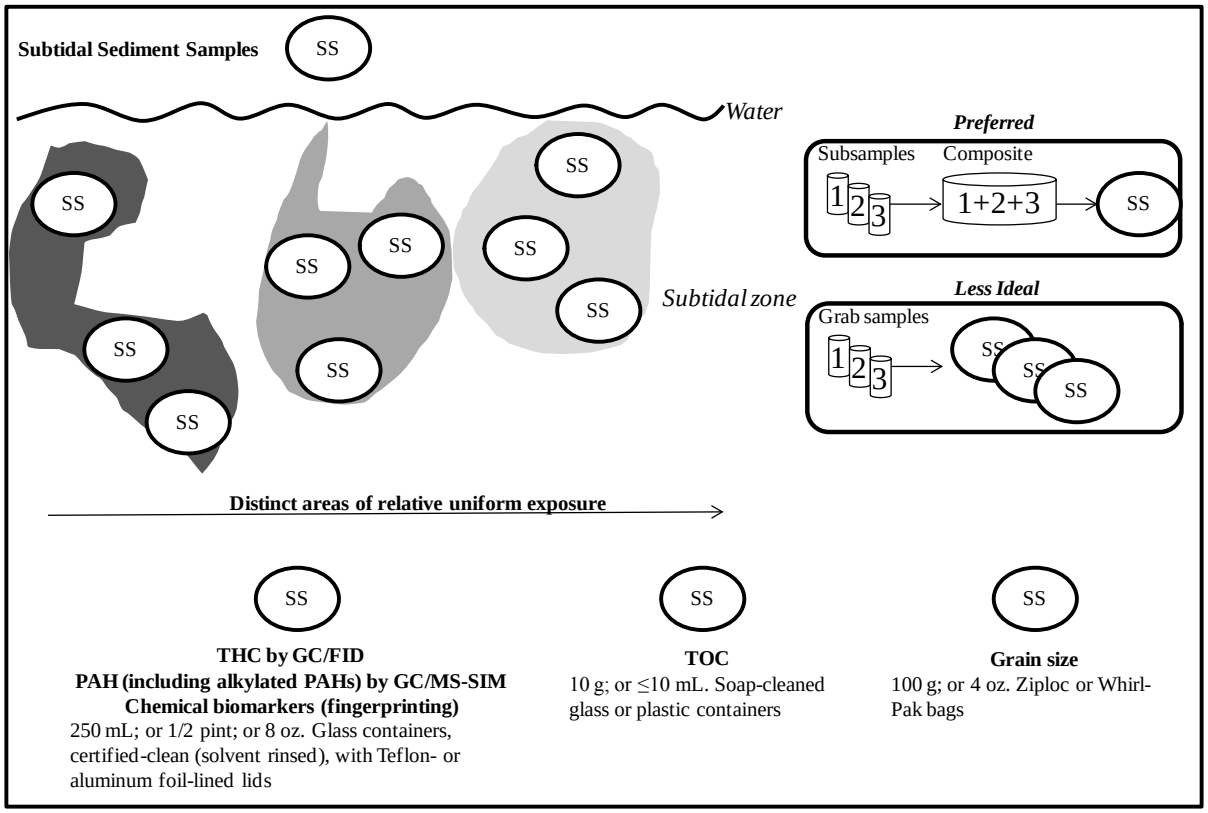

Figure 1. Schematic representation of the recommended subtidal sediment sampling strategy. SS= subtidal sample. Area-specific modifications may be needed.

- For each site, record:
  - Date, time, weather conditions (e.g., wind direction and speed)
  - Physical setting of adjacent habitats (shoreline orientation, exposure, etc.)
  - Depth of subtidal sediments (in meters)
  - Sediment type
  - Presence of snow or ice along shoreline or in the water
  - Presence of biological resources or other relevant information
- Determine the number and location of samples, as follows:
  - Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area

  - Minimum guidelines under normal conditions are at least three samples per location of relatively uniform oiling exposure (e.g., three samples within 500 m of the shoreline in a small bay, or three samples in a small lagoon)
  - If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiled location, and two heavily oiled locations
- If specified in the work plan, collect separate splits of homogenized samples for infauna or toxicity testing, so they can be correlated with chemical results.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site using to sample collection to document the site conditions, as well as the sample collected. If possible, make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline). If taking photographs of the sampling site is impractical, only take photographs of the sample collected.
- Any sediment sampling device which meets the following requirements can be used:
  - Creates a minimum bow wake when descending
  - Penetrates the sediments to below the desired sampling depth
  - Closes to form a leak-proof seal after the device is triggered to close
  - Prevents sediment washout and disturbance when ascending
- Use dredges for hard substrates and coring devices in sedimentary substrates and inundated tundra, unless water depth precludes effective sample collection.
- Clear surface slicks prior to deploying the equipment by sweeping the area with a sorbent pad or placing a barrier up-current to divert surface oil around the sampler deployment area.
- When deploying and retrieving the sampling device:
  - Lower and retrieve the sampling device at a controlled speed of ~30 cm per second to minimize potential bow wake activity and bottom disturbance as the sampler contacts the bottom, and loss and disturbance of the subtidal sediment sample during retrieval
  - The device should contact the bottom gently, making sure it settles flat; only its weight or piston mechanism should be used to penetrate the sediment. It is important to minimize disturbance to the surface floc, which is likely to contain oil contaminants if they are present
  - Secure the samplers on board and examine the sample for acceptability based on the following criteria:
    - The sampler is not overfilled; the sediment surface is not pressed against the sampler top
    - Overlying water is present, indicating minimal leakage
    - Sediment surface is undisturbed, indicating lack of channeling or sample washout
    - The desired penetration depth is achieved (e.g., 4-5 cm for a 2 cm sample)
  - Siphon off the overlying water near one side of the sampler
  - Using a flat scoop, accurately collect the top 2 cm from the center of each grab, avoiding sediments in contact with the sides of the sampler. Collect other intervals, as needed, using a new scoop for each sampling interval

  - Take composite samples of at least three subsamples within the same dredged material (collected from the same site) and sampling interval, and homogenize composite sample before placing in the jar(s) using a disposable aluminum pan or aluminum foil-lined container
  - If time, equipment, or logistics do not allow collection of composite samples, collect grab sediment by following steps above, omitting the collection and mixing of subsamples. Place grab samples directly into jars for mixing, homogenizing and splitting in the laboratory
  - In addition to collecting samples for chemical analysis, take samples for TOC (placed in soap-cleaned glass or plastic containers) and grain size (placed in Ziploc or Whirl-Pak bags) analysis

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines), noting where each subtidal sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (sediment)
  - Sample #, date/time
  - Sampling method (grab, core, composite) and sample collection depth
  - Approximate distance from shoreline
  - Note if sample is for QA/QC (field blank, trip blank, rinsate blank) or if it is a spilt or duplicate sample
  - Sediment oiling conditions (using standard shoreline assessment terminology), water depth, weather conditions (e.g., wind direction and speed), sediment characteristics (see next bullet), presence of biota, vegetation or debris, odors, and other relevant information on the field data sheet
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.; vertical changes in sediment characteristics
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- If sample volume is split between two jars, both jars should receive the same sample ID and be recorded on a single line of the Chain of Custody form.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately place all sediment samples for chemical analysis in a cooler and keep at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature if heated storage space is not available. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Refrigerate (do not freeze) samples for TOC. Samples for grain size do not require refrigeration.
- Protect samples for chemical analysis from direct sun exposure (e.g., UV radiation).

- Samples for TOC and grain size can be stored separately from samples for chemical analysis.
- Tape lids on sample bottles so that they do not accidentally come off.
- If possible, store samples for chemical analysis from unoiled areas in one set of coolers, with oiled samples in a separate set of coolers.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping. Take special care with gravel sediments because individual pieces can rattle around during shipping and break the glass jars.
- Freeze samples for chemical analysis as soon as practical or by the end of each day if samples are not going to be analyzed within 7 days of collection.
- Sediment samples can be held frozen in the dark for several years without loss of sample integrity.
- Sediment extracts can be held at 4°C in the dark for 40 days without loss of sample integrity.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.
- Ship to the laboratory as soon as practical with completed Chain of Custody (COC) forms.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels [a]	Recommended Holding Time	Minimum No. of Samples per Location
Total Hydrocarbons (THC) by GC/FID	250 mL; or 1/2 pint; or 8 oz. Filled ¾ full [d]	15 µg/kg	7 days at 4°C [b] or several years frozen [c]	3
PAH (including alkylated PAHs) by GC/MS-SIM		0.001-0.01 µg/kg		
Petroleum biomarkers (fingerprinting)		0.001-0.01 µg/kg		
Total Organic Carbon (TOC)	10 g; or ≤10 mL. Filled ¾ full	0.01%	28 days	3
Grain size	100 g; or 4 oz	NA	NA	3

[a] µg/L= ppb; [b] Store at 4°C in the dark; [c] Stored frozen in the dark; [d] Several analyses can be made from a single sample.

Analytical Methods

- **Total hydrocarbons** (THC). Often referred to as total petroleum hydrocarbons (TPH), but most methods do not differentiate among petroleum, petrogenic, and biogenic hydrocarbons. THC by GC-FID (total area of FID gas chromatogram of combined f_1 and f_2 fractions after column chromatography; e.g., EPA Method 8015 Modified) is often the preferred method because of the low detection limit (compared to other THC methods) and the direct measurement of hydrocarbons. This method does not detect low boiling point compounds (below n-C_8). THC analyses generally will not provide the data needed to support calculation of toxic effects from PAH exposure, and will have to be corrected to equivalent PAHs. The THC results, however, can be used to document changes in oil weathering and map extent of exposure of water column resources, if meaningful detection limits are used. THC results can be used as a screening tool to estimate the presence and amount of

hydrocarbons and provide an indication of which samples should receive highest priority for more extensive analyses.

- **Polynuclear aromatic hydrocarbons** (PAH). Since most of the toxicity in oil is due to the PAHs, it is often the preferred analysis. However, PAHs are expensive and require special laboratory skills. If PAHs are to be measured, it is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard PAH "priority pollutants". This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs, using GC/MS in the selected ion monitoring mode (SIM). Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. Have the lab also run the source oil.
- **Petroleum biomarkers**. These chemicals are the most important hydrocarbon groups used for chemical fingerprinting allowing a quantitative identification of the source oil. Because biomarkers are more resistant to weathering and biodegradation than other hydrocarbons, these can also be used to quantify the degree of oil weathering. Biomarkers include steranes/triterpanes, which are "fossil" compounds unique to the oil formation and compounds that provide a secondary and confirming line of evidence in forensic oil identification. Chemical biomarkers are typically analyzed by GC-FID (e.g., EPA Method 8015 Modified). These chemicals are typically analyzed concurrently with THCs
- Analyses may also include:
  - **Sediment grain size**. There are different methods for measuring sediment grain size, and may include sieve analysis of gravel to sand fractions, pipette analysis for muddy sediments, or more sophisticated methods that use rapid sediment analyzer equipment
  - **Water content**. Water content is typically measured in conjunction with chemical and TOC analyses by drying a specified mass of sediment in an oven at constant temperature (typically 100-110 °C) and calculating the mass difference between wet and dry sediments
  - **Total organic carbon**. There are different methods for measuring TOC in sediments, and may include high-temperature combustion methods or more sophisticated methods that use total organic carbon analyzers

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Appendix A
Supporting Documentation- Field Data Forms

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Soil/Sediment Sample Collection Form

Sample Collection Form - SOIL/SEDIMENT

Lead Sampler's Name/Phone		**Sampler Team Code**	
Lead Sampler's Affiliation		**Resource Group**	
NRDA Contact/Phone		**Resource Group Leader**	
Incident Name		**Habitat (e.g., sand beach)**	
General Location Description		**Sample date (mm/dd/yyyy)**	

Location Code	Matrix	Sample Number (two digits)	Sample Time	Sampling Method	Sample Collection Depth	Depth units	Sample QA/QC Type	Latitude	Longitude	Sample Notes
NRDA Sample Grid ID	*(S)ediment or Soil (L)*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Method of sampling (i.e., core, grab or composite)*	*Include upper and lower collection depth. Use 0 for surface samples.*	*Units for depth values*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD - YYY.YYYYYY*	*Description of sample, equipment used, including estimated volume, photo numbers, etc.*

Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)

Samples Relinquished by:				Received by:			
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment	(GR)ab	(c)m	
Soil (L)	(CO)re	(m)	
Blan(K) Water		(i)nches	
		(f)eet	
Oil, Tarball or Water	**Sampling Method**	**Sample Position/Depth**	
(O)il	(GR)ab	(FLOAT)ing	
Tarball (B)	(SC)rape	(SUB)merged	
(W)ater	(OT)her	(STRAND)ed	
Blan(K) Water		(COV)ering	
Other (H)		0 - (Surf)ace	
		<depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue	(WH)ole body	(MU)scle	
Wrack (R)	Whole body w/o shell (WNS)	Yolk	
Blan(K) Water	Chorioallantoic Membrane (CAM)	NA <for Wrack only>	
	Egg (EM)bryo		
	Fillet with skin (FS)	**Species**	
	Fillet without skin (FWOS)	<enter species>	
	Gall Bladder (GB)	NA <for Wrack only>	
	Leaves (LEV)		
	Leaves and stems (LVS)		
	(LI)ver		
Sample Identifier system			
Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			
QA/QC types:		Other sample types:	
Field Blank (FB)	Rinsate Blank (RB)	(S)plit	
Trip Blank (TB)	(D)uplicate		

Guidelines for Collecting Ephemeral Data in the Arctic: SHELLFISH TISSUES

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting shellfish tissue samples from intertidal or subtidal areas during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Bivalves are the most commonly sampled shellfish for chemical exposure analysis, but many other invertebrates, including other mollusks and crustaceans can be sampled using this guideline.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in the biological tissues compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate

Study exposure and injury

- Document the extent and duration of exposure to the spilled material
- Document the bioavailability and exposure pathways of the spilled material
- Quantify oil chemicals in shellfish tissues
- Document routes of exposure for higher trophic level organisms
- Document morphometrics or other biological parameters for injury assessment

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of biological characterizations

Collaboration

- Support other ongoing efforts including, but not limited to, modeling of oil transport, exposure and impacts to water-column and benthic resources (see Water, Snow, and Sediment guidelines), and assessing the risk to higher trophic organisms consuming contaminated prey

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as a rocky shore or lagoon
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity. Generate a list of alternate sites that are prioritized to fall back on when unforeseen circumstances prevent primary site sampling.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guidelines may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- A stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected, is recommended if no other sampling strategy has been developed. This type of sampling improves the representative quality of samples by reducing sampling error (variability).
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with shellfish tissue sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); pre-cleaning aluminum foil for sample storage, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.

- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled and unoiled reference locations.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what locations have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize site selection. In this case, highest priority samples are to be collected from oiled areas that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiled shellfish samples from sensitive/productive sites that are likely to be oiled is also a priority. Sampling at unoiled "control" sites and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Consider the availability of historic or baseline data, such as NOAA National Status and Trends and mussel watch data, when choosing sampling areas. Sampling at or near sites for which baseline data is available should be prioritized.
- Shellfish tissue samples should be collected pre-oiling, if possible, as soon as practical after oiling and periodically thereafter.
- A power analysis may be needed (if relevant information is available) to determine the number of samples needed to detect minimum statistically significant differences among locations:
  - These calculations should be performed by a trained statistician
  - Use all available existing data or alternative data from similar field studies, with information from background/reference locations, and from locations with different levels of oil contamination. NOAA National Status and Trends, EPA EMAP, or state Mussel Watch programs may have background data for contaminants in shellfish and sampling protocols
  - Types of data used in these analyses may include: tissue concentrations, gonad size, tissue weight
  - Data needed include: average and standard deviation values per location, a minimum desired change of environmental or biological significance (example: 2x tissue concentrations above background, 0.1x gonad size reduction relative to background/reference sites)
  - Estimate the number of samples required to obtain a desired power (typically, values between 0.8 and 0.95 at α=0.05) using publically available power-calculator tools (example: http://www.psycho.uni-duesseldorf.de/abteilungen/aap/gpower3/). By definition, high power decreases the chances of failing to reject a false null hypothesis or making a Type II error (concluding that there is no statistically significant difference when in fact there was one)

- Use a computer or conceptual model of the extent of habitat contamination or an appropriate power analysis (see above) to determine the number of locations and sites to be sampled.
  - Minimum guidelines are at least 3-5 samples per location of relatively uniform exposure or distinct waterbody
  - If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiled location, and two heavily oiled locations
- Choice of species is important. Prior to sampling, review any existing literature on shellfish in the study area, and if available, choose a species already studied with regards to PAH exposure. When selecting species for sampling, consider ecological characteristics including trophic guild (e.g., filter feeders, grazers, predators, etc.), life history strategy, etc.
- For exposure characterization, consider targeting species with wide distribution in the study area and that are readily available across all locations and in quantities high enough for repeated sampling over time. For ingestion risk assessment, target key food species. In the Arctic, with a narrow tidal range and soft sediments, there are little to no shellfish in intertidal areas. Shellfish in the Arctic include crabs, clams (razor, butter, pinkneck, and softshell) and gastropods (moon snails, neptune snails and others) which are present in nearshore and offshore areas. Other invertebrates, such as isopods and amphipods may be available for sampling as well, but their life histories differ significantly from bivalves and gastropods. Shellfish increase in abundance in intertidal areas further south, including mussels attached to hard surfaces.
- Temperature can have a large impact on shellfish physiology. Some animals stop feeding or even passing water over their gills at low or high temperatures. Be aware of these differences when selecting species for monitoring and comparing results among species and over time.
- Uptake and depuration rates vary widely among species. Depuration usually takes weeks; thus shellfish sampling should be initiated within 1-2 weeks after maximum exposure.
- Procedures may need to be modified/adapted to collect tissues for species with unique biological or ecological characteristics.
- Sample along exposure gradients, starting in the cleanest zone and at regular intervals proportional to the exposure area.
- Shellfish samples can be collected from boats, the shoreline, or by wading in shallow water.

Collaboration
- Shellfish tissue samples can be collected in conjunction with water and sediment sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the study plan, desired sampling capacity, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.
- Coolers – for sample storage and transport
- Ice packs/collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves (preferred) when sampling for shellfish under extreme cold conditions
- Sampling jars – 4 or 8 oz certified organic-clean jars with Teflon-lined lids and labels:
- Aluminum foil: the dull side should be pre-cleaned with acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) to wrap shellfish samples, with the dull side in contact with the sample

- Ziploc or Whirl-Pak bags
- Dredges, knifes, tongs, trowels, grabs– washed and solvent cleaned, wrapped in foil and sealed for transport − for shellfish collection
- Shovel
- Rake
- Sieve with 500 mm mesh
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Field notebook, evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries)
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment are not available):

- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel knives).

- The only equipment to be used between sites are a shovel and a dredge, which should be cleaned with soap and clean water. Repeated digging in clean sediments can be a last resort for cleaning the shovel if soap or clean water are not available. Alternatively, use a clean dry towel or other dry material to clean the shovel before its next use. Additional cleaning may be required when working at oiled sites (see below).
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples, including dredges:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on boots, shovels and other equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.
- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- Collect primarily live animals (shells intact and tightly closed). If live animals are not available, collect dead animals only if tissues appear to be fresh. Note the collection of live/dead animals on the field sample forms.
- If large shellfish die-offs are observed, note the location, species, life stage, visible oiling and approximate number or extent of the mortality event. Take pictures to document the die-off. Consider collecting samples to determine cause of death or for chemical analyses.
- Attached organisms are pried away from the substrate with a knife, trowel, etc.
- A dredge that is dragged through the substrate may be needed to sample shellfish in subtidal habitats. Dredges that are inserted into the sediment at a point location may also be used but are unlikely to obtain a sufficient number of individuals for analysis.
  - Deploy dredge from the side of the vessel that offers more space for safe deployment and retrieval, and that is further away from sources of contamination (e.g., fumes)

  - Record the GPS coordinates of the start/stop positions (when the dredge enters and the dredge and before the dredge is brought onboard)
  - Dredge the sediment surface for 3 minutes at 2 knots in a circular pattern. Repeat this step at least 3 times per site (3 replicates), or until the desired target number of individuals is obtained
  - Follow cleaning and storing procedures described elsewhere
  - All individuals obtained via dredging MUST be collected during the same sampling event (2 hour sampling maximum)
- Infaunal samples should be rinsed with site water (clean water, if possible) to remove sediments except when oil sheens and slicks are present.
- Shellfish may also be collected using traps (mobile crustaceans) (see photograph) and beam trawls (epibenthic invertebrates). Seining and fyke nets may also produce shellfish in some areas.
- Shellfish samples can be collected for different purposes:
  - Composite samples (preferred) by species for body burden:
    - Familiarize yourself with the target species and ensure that accurate taxonomic identification has been made during sample collection such that there is no mixing of closely related species. Research the literature for the average wet weight per size of the species to be collected. For example, to meet the target of 30 g wet weight of tissue, a sample would need to include 20 blue mussels 3-5 cm in length or 15 littleneck clams 3 cm in length. If uncertain about the number of individuals needed to meet minimum weight requirements (30 g wet weight), open, shuck, and weigh individuals of a certain size for calibration

    - Samples should be collected along the same vertical horizon or water depth, and from the same oiling exposure
    - Collect enough tissue weight to meet detection limit objectives and to average out the variations at a site among individual organisms:
      - Take at least 3-5 composite samples per site of relatively uniform exposure or distinct waterbody, each composite sample having approximately 30 g wet tissue weight,
      - On shorelines, take individual organisms for composite samples within a 5 m radius of each other, or within the closest area possible, collecting individuals of the same size. As a general rule, the smallest individual in a composite sample should not be <80% smaller than the largest individual
      - All individual organisms used in a composite sample MUST be collected during the same sampling event (2 hour sampling maximum)
    - To minimize the risk of contamination, do not open shellfish in the field; collect the entire animal and shell
    - Wipe oiled shells with sorbent pads, wipes, etc. If heavily oiled, use a solvent damp wipe
    - Samples should be individually wrapped on a double layer of heavy duty foil and placed into certified-clean glass jar (preferred) or directly into a double Ziploc-bag (less ideal), or placed directly into certified-clean glass jar (least ideal). For bags, label the inner bag with marker pen and a waterproof sample label placed between the two bags. Jars are labeled with an adhesive label directly placed on the lid. Use clear tape to protect the paper label. Make sure all organisms within the composite sample are stored together

        - Immediately place samples in coolers on ice
        - If time is a concern, the primary target size range should include the larger individuals harvested at each sampling site
        - All individual organisms used in a composite sample MUST be collected during the same sampling event so that temporal changes in contaminant concentrations are minimized
        - Follow the sample cleaning and storage guideline provided above
    - Fingerprint analysis:
        - Individual size is not important so long as the required mass is collected (approximately 30 g wet tissue weight). However, if space in coolers is limited, try to collect specimens in the upper end of the species' size range
        - Collect a sufficient number of organisms to satisfy the mass required for these analyses. A larger number may be needed depending on the need for performing analyses on specific organs (gut)
        - Follow the sample cleaning and storage guideline provided above
    - For other objectives (e.g., morphometrics, gonad assessments, or other biological parameters):
        - Individuals may need to be of the same shell (or body) size
        - Collect at least 10 individuals per species per site for gonodal condition and other health parameters. The same individuals can be used for morphometric analyses. If space in coolers is limited, these organisms can also be used for lipid and water content analysis (less ideal). Alternatively, collect an additional 5-10 individuals for water content analyses
        - Samples collected for analyses of gonodal condition and other health parameters MUST be processed within 72 hours of collection
        - Measure shell size range of collected shellfish
        - Follow the sample cleaning and storage guideline provided above
- Record the presence of oil, weather conditions, etc. in field notes.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each sheen sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
    - Sample collection site (NRDA sample grid ID and GPS coordinates)
    - Sample matrix (shellfish tissue)
    - Sample #, date/time, site location, tidal elevation, water depth
    - Species collected, type (live/dead), number of individuals, size range, sample type (whole, tissue only)
    - Describe the oiling conditions (using standard shoreline assessment terminology), weather conditions (e.g., wind direction and speed), sediment characteristics, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
    - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.; vertical changes in sediment characteristics
    - Record observations of any external evidence of contamination

- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil exposure is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oiled samples separate from lightly or unoiled samples to reduce risk of cross-contamination.
- Samples should be received by the laboratory for processing within 7 days of collection.
- Immediately place all samples in cooler and keep at 4°C. Freeze samples for chemical analysis as soon as practical.
- Depending on the remoteness of the sampling location, laboratory processing may not be achievable. If this or any other issues are anticipated, freeze samples to -20ºC as soon as practical and ship samples to the laboratory following special shipping required to maintain samples in a frozen state. Once properly frozen, they can be held for years without loss of sample integrity.
- DO NOT freeze samples collected for analyses of gonodal condition and other health parameters. These can be kept at 4°C until processed.
- Tape lids on sample bottles so that they do not accidentally come off.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical, overnight (preferred), with completed Chain of Custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed, and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Size

- For PAH analysis by GC/MS-SIM the minimum sample size is 30 g wet weight (considering soft tissues only, not shells) (composite of individuals of similar size).
- For analysis of lipid, water content, etc. the minimum sample size is 5-10 individuals (depending on size).

Analytical Methods

- **Polycyclic aromatic hydrocarbons** (PAH). Because most of the toxicity in oil is due to the PAHs, it is often the preferred analysis. However, PAHs are expensive and require special laboratory skills. If PAHs are to be measured, it is important that the analytes include the alkyl-substituted PAH homologs, in addition to the standard PAH “priority pollutants”. This method is referred to as Modified EPA Method 8270, because the list of PAHs is expanded to include the alkylated homologs (approximately 43 PAHs), using GC/MS in the selected ion monitoring (SIM) mode. Detection levels should be at least 0.1 ppb for individual PAHs to support injury assessment using toxicity thresholds. Have the lab also run the source oil with each batch.

- **Lipid Content**. Lipid content is defined as the percent of sample tissue extracted and remaining after solvent evaporation using dichloromethane. It is used to normalize organic contaminants in tissues, to aid in spatial and temporal comparisons across samples.
- **Water Content**. Most results are reported as dry weight, to reduce sample variability. Laboratory should report wet and dry weight, and water content.

Key References

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Appendix A
Supporting Documentation- Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:
- Tissue/Wrack Field Data Form

Sample Collection Form - TISSUE/WRACK										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Species (NA for Wrack)**	**Tissue Type (NA for Wrack)**	**Number in sample (NA for Wrack)**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(T)issue or Wrack (R)*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Species collected*	*Whole or tissue type*	*Number of organisms in sample*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD - YY'Y.YYYYYY*	*Description of sample, including size (weight, length), equipment, photos numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										
Sample relinquished by:				**Received by:**						
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**			

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball or Water	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H)	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

EPHEMERAL DATA COLLECTION GUIDELINES:

HABITATS AND ASSOCIATED COMMUNITIES

Guidelines for Collecting Ephemeral Data in the Arctic: SAND BEACH AND TIDAL FLAT INFAUNA

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guidelines Objectives

The primary objective of this document is to provide guidelines on collecting infauna samples from sand beaches and tidal flats during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in sand beach and tidal flat habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate

Describe habitat

- Estimate the areal extent and degree of oiling in sand beach and tidal flat habitats
- Document the presence/absence and species composition, and estimate the abundance or density of sand beach and tidal flat infauna

Study exposure

- Document the extent and duration of exposure to the spilled material and its bioavailability
- Measure oil-related compounds in biological tissues
- Support exposure and transport modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical and biological characterizations

Collaboration

- Support other ongoing efforts (see Intertidal and Subtidal Sediment guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Sand beach/tidal flat infauna are difficult to sample because of their inherent heterogeneity over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as a barrier island or lagoon
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, etc. that may compromise sample integrity.
- Consult ESI maps, state resource guides and other resources to determine what infauna may be present in the sampling area. Adjust the sampling strategy accordingly to target key species of interest.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of sand beach/tidal flat contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per site needed to respond to the sampling objectives.
- A stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected, is recommended if no other sampling strategy has been developed. This type of sampling improves the representative quality of samples by reducing sampling error (variability).
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling transects, transect spacing, sampling zone width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with sand beach/tidal flat infauna sampling. Tarballs, sheens, or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars and aluminum foil for sample storage, etc.

- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what locations have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled sand beach and tidal flat areas that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiled samples from sensitive/productive areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Infauna samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency is a function of oil persistence, biological community, habitat importance, and resource availability and should be defined in the study design.
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting sand beach and tidal flat infauna is at least three samples per tidal zone per location of relatively uniform oiling exposure. If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiling location and two heavily oiled locations.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration

- Infauna samples can be collected in conjunction with intertidal and subtidal sediment sampling, nearshore water sampling, and stranded oil sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts are important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for infauna sampling under extreme cold conditions
- Sampling jars – 4 or 8 oz certified organic-clean jars with Teflon-lined lids and labels
- Surveying supplies – site markers (appropriate for substrate type), surveying flags and tape, quadrats (0.25 m^2 and 1.0 m^2), 30 m fiberglass tape measure marked in cm, hand counter, field balance, caliper, box screen with 5 mm mesh, sieve with 0.5 mm mesh, hand coring device (cylindrical, 0.01 m^2 or similar size), rubber stopper for coring device
- Pencils, waterproof pens, waterproof labels, markers
- Large tub or bucket
- Pre-cleaned aluminum foil
- Sample bags/jars
- Shovel
- 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal) – for sample preservation; and a stain (such as Rose Bengal) if appropriate
- Field Sample forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Shoreline terminology code list and shoreline assessment survey guidelines (see Stranded Oil guidelines)
- Field sample forms (sand beach fauna identification field guides/charts), field notebook (waterproof paper), random number table sheets, shoreline oil terminology code list, other guidelines as needed (Field Photography, Subtidal and Intertidal Sediment guidelines)
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment is not available):

- Sufficient quantities of pre-cleaned or disposable, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents

- Teflon solvent squirt bottles
- Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
- Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- The only equipment to be used between sites is a shovel, which should be cleaned with soap and clean water. Alternatively, use a clean dry towel or other dry material to clean the shovel before its next use. Repeated digging in clean sediments can be a last resort for cleaning the shovel. Additional cleaning may be required when working at oiled sites (see below).
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples collected for chemical analysis:
  - Wash sampling equipment with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on boots, shovels, and other equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Study Design Implementation

- If recent SCAT data are available, it may not be necessary to conduct an overflight or ground survey. Otherwise, if practical, conduct an overflight of the entire affected area within two hours of low tide

(before or after to locate sand beaches and tidal flats and observe the extent of visible contamination). Note that weather, particularly fog, may dictate survey times that may not coincide with tides. Tides may not be a consideration along high Arctic shorelines where tidal range is small and water level is primarily wind driven.

- If an aerial survey is not feasible, survey from the ground. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs can be geo-referenced later. Observations should include:
  - Locations and approximate lengths of sand beaches and tidal flats in the oiled area
  - Degree of oiling of these habitats (use standard SCAT terminology; see Stranded Oil guideline)
  - Locations of access points, major landmarks, and potential ground-truth and reference stations
- Consider the accessibility of the locations for several methods of access (e.g., plane, helicopter, boat, foot, etc.). It may not be possible to safely access all locations of interest for sampling.
- Mud and glacial silt flats present unique safety risks - people and equipment can become stuck.
- At each selected sampling location, conduct a preliminary visual survey of the shoreline and draw a field sketch showing:
  - Shoreline orientation, type, grain size, etc.
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
- At each selected location, establish at least three transects at least 30 m spacing apart (Figure 1).

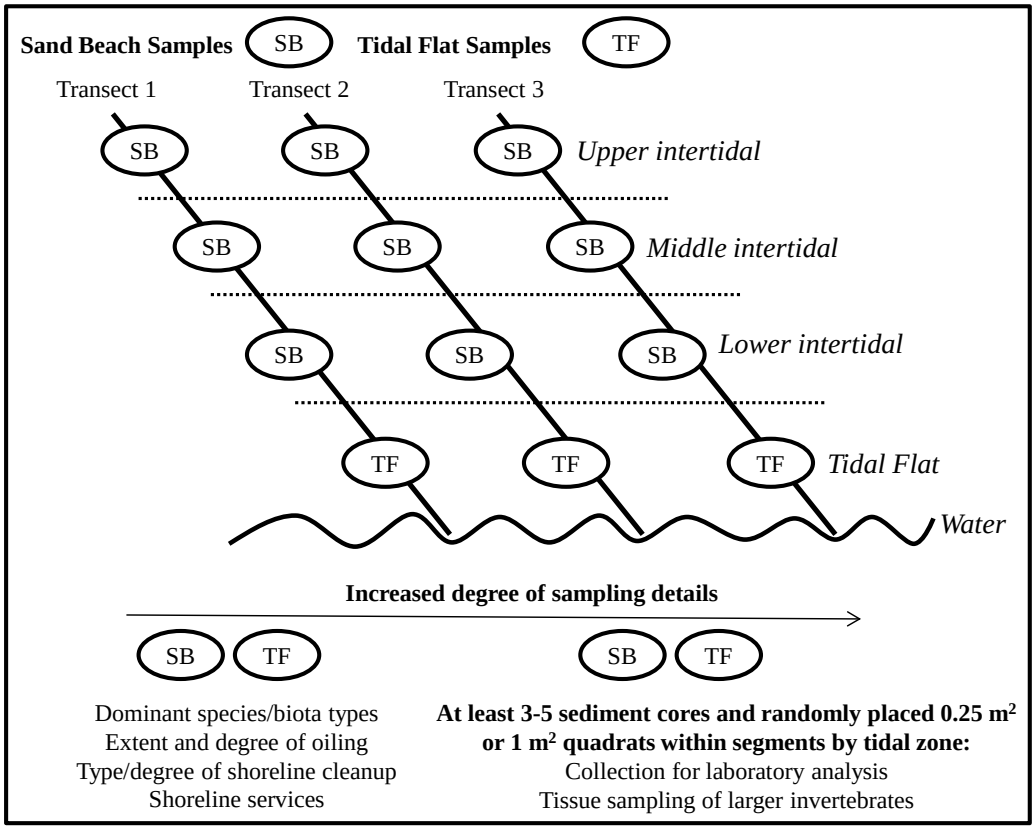

Figure 1. Schematic representation of the recommended sand beach/tidal flat sampling strategy, including transects and sampling sites. Dashed lines represent approximate tidal zones. SB= sand beach, TF= tidal flat. Area-specific modifications may be needed. For example at locations with very narrow intertidal zones, only one or two sampling intervals may be used.

- If no other sampling strategy has been developed, use a stratified random sampling approach by randomly selecting the first transect starting point and expanding the sampling site (other transects) systematically into a grid from that initial point. Randomly select sampling sites within each intertidal zone intervals.
- When establishing ground transects:
  - Record transect location using a GPS, but also accurately plot the transect on a map or aerial photograph (if available)
  - Permanently mark transect locations using "front and back" stakes that line up along the transect lines, and consider stake placement carefully to minimize loss to vandalism, erosion, ice-scouring, etc. Label the stakes to clearly identify the site as a NRDA sampling location. Assure that stakes do not present a hazard to people traveling by ATV or snow machine
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one stake is lost; note whether the angle reading is magnetic or corrected to true north
  - Take photographs of the transect at the beginning, middle and end, including upslope, downslope, and longshore images. This takes little time and establishes a reference for future work
- If possible, run transects so that the lower intertidal area can be sampled within two hours of low tide (before or after). In the Chukchi and Beaufort Seas the tide range is limited (<30 cm); wind-driven storm surge is the most determinant factor in water height and should be considered when sampling. Tides are only a consideration south of the Bering Strait where the tidal range is >1 m. Transects should be run perpendicular to the shoreline and encompassing the entire intertidal zone, including the supratidal zone (e.g., first barrier or the vegetation line). This is important because in the Arctic storms are likely to push oil into the suptratidal zone. If feasible, include the lowest accessible tidal flat zone.
- If time/tide constraints exist, focus on sampling the lower and middle intertidal, and the upper edge of the tidal flat.
- On some high-Arctic shorelines, the intertidal zone may be very narrow. If this is the case, two sampling intervals can be defined on each transect, in the upper (or storm surge) and lower intertidal zone, or just one station if the intertidal zone is very narrow.
- Along each transect, use standard oiling terminology codes and estimation charts to record in field sample forms or field notebooks:
  - Date, time, weather conditions (e.g., wind direction and speed), tide level (as observed), and initials of observers
  - Distance of the interval
  - Physical setting (shoreline orientation, exposure to wave energy and tidal currents, etc.)
  - Length of the transect (in meters) and of the sampling zone
  - Sediment type and grain-size (e.g. mud, sand, mixed sand and gravel, etc.)
  - Dominant species or types of biota present (including signs of infaunal organisms such as burrows, worm tubes, fecal mounds, etc.)
  - Presence, condition, and/or altered behavior of visible biota such as amphipods, gastropods, crabs, etc.
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts; see Stranded Oil guideline). Be sure to record depth of oil penetration into the sediments, if any (using a shovel or coring device)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Presence of snow or ice along shoreline
  - Presence of biological resources and other relevant information
- Collect samples from each intertidal zone (described below) and record the distance along the transect and GPS coordinates of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If time allows a calibration exercise is recommended prior to field sampling to ensure that all field teams consistently perform infauna sampling.
- If sampling sites are covered by snow or ice, carefully remove the snow/ice without mixing or disturbing the sediment underneath and proceed with infauna sampling. Note snow and ice conditions on the field data sheet. If snow or ice present in the intertidal zone are impacted by oil (as opposed to deposited on top of oiled sediments), it may be desirable to collect snow/ice samples for chemical analysis (see Snow guideline).
- Carefully remove stones, sticks, and other debris on the sediment without mixing or disturbing the sediment underneath.
- Sampling can be conducted to document the presence, composition, and general abundance of organisms. In the Arctic, sand beach and tidal flat infauna may include amphipods, bivalves, polychaetes, and oligochaetes.
- If highly quantitative density estimates are needed, or detailed comparisons of oiled vs. un-oiled locations are planned, an experienced sand beach ecologist and statistician should be consulted to plan more detailed studies. If one is not available, the following sampling should be performed.
- For semi-quantitative surveys of infauna:
  - For species presence/absence, composition, or density estimates, take 3 replicate sediment cores at each tidal elevations along each shoreline transect
  - Cores should be taken within 5 m of the transect line
  - Photo-document the sampling locations, sample collection methods, organisms observed, any obvious oil impacts, etc.
- To sample macroinfauna, use a hand coring device, typically a cylinder with an open cross-sectional area of 0.01 m^2 and a small hole on the top:
  - Record the distance along the transect and GPS coordinates of each sediment core location
  - Using a ruler, mark the outside of the corer every ½ cm so that the depth of each core sample can be measured and recorded. Cores should not exceed 15 cm in depth
  - Prior to collecting cores, carefully remove debris, shells, and other material without disturbing the sediments
  - Take photographs of each sediment core location
  - Insert the corer into the sediment (in a vertical position) to a maximum depth of 15 cm
  - Place a finger or thumb over the small hole (or insert a rubber stopper) to create a vacuum and extract the core
  - In coarse-grained sediments, cores often cannot be extracted whole, and a metal plate or other similar object must be slid under and across the bottom of the core before it is removed
  - If field sieving of cores is not possible, empty the contents of the core into a labeled container, such as a large plastic bag. Preserve with 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate

- Core samples can be sieved in the field (using a 0.5 mm screen). When sieving, gently force water up through the bottom of the sieve, by bobbing the sieve up and down in a large bucket or tub of water. This prevents forcing animals into or through the bottom of the sieve
- After sieving, place samples in a sample bag or jar and preserve with 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate
- If field sieving of cores is not possible, preserved whole core contents in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal)
- Place a waterproof label with the location, sample number, and date inside the sample container
- To avoid cross contamination, make sure sediment cores are cleaned between samples

- To sample infaunal organisms for chemical analysis, follow Shellfish Tissue guideline. Small infaunal organisms of the same species can be composited to achieve desired sample mass.
- To sample larger macroinfaunal target species, such as bivalves harvested for commercial or recreational purposes:
  - Select a sampling site at random by placing a 0.25 m^2 or 1.0 m^2 quadrat on the sediment surface. Quadrats should be placed within 5 m of the transect line. One approach to random sampling is as follows:
    - Place two tape measures at a 90°angle along two sides of the sampling location
    - Use a random numbers table to randomly select two numbers
    - Place the center of the quadrat at the intersection of the two numbers on the tape measure
  - Record the GPS coordinates of each quadrat
  - Take high-resolution photographs of the entire quadrat from an angle as vertical as practical. This will facilitate computer-based analyses of individual quadrats. When taking photographs observe the following:
    - If the presence of large or abundant motile invertebrates block the view of sessile invertebrates, carefully remove these organisms from the quadrats prior to taking photographs and without disturbing less motile invertebrates, but make sure to document the removal of organisms in the field sample form
    - High-resolution photographs must include all four sides of the quadrat as these will be used to digitally count individuals and measure their coverage on a computer screen
    - Quadrat frames can be split into 2-sided frames to facilitate computer-based analyses
    - Photographs need to be relatively flat so that the entire quadrat falls within a similar focal plane, with minimal shadowing from crevices or projections
    - Ideally, photographs should be taken during the lowest tide and best light conditions (e.g., closest to midday or when overcast). Avoid shooting into the sun and avoid including sky, ocean, or tidepools in the view
    - If possible, use a quadrapod apparatus to support the camera at a constant height (1 m with a 35 mm lens) from the quadrat and position it to capture all four corners of the quadrat:
      - A quadrapod consists of a gray PVC pipe frame with a photoplot-size bottom (0.5 m^2 or 1.0 m^2 internal dimensions) connected by 4 poles to the frame supporting the digital camera
      - Strobes mounted laterally and away from the camera can enhance lighting on the quadrats and reduce shadows
    - The best quality photographs are obtained by optimizing the ISO, aperture, and shutter speed
    - Remember that all quadrat images must be of sufficient quality to allow a positive identification and enumeration of the species in the quadrats
  - Prior to removing quadrat contents, carefully remove debris, shells, and other material without disturbing the sediments

  - Use a shovel to excavate the sediments within the quadrat, commonly 30 cm. It may be necessary to excavate deeper if certain species, such as clams, are present (consult ESI maps and other resources for species information for sampling locations)
  - Sieve the excavated sediments using a 5 mm screen (or larger, depending on the size range of the target species)
  - Identify, count, weigh, and/or size the captured organisms in the field, and release them live, or, place samples in labeled plastic bags or jars and freeze, or preserve in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate
  - Place a waterproof label with the location, sample number, and date inside the sample container
- If chemical analyses of tissue are planned for larger infauna, such as bivalves, follow the Shellfish Tissue guideline. Briefly:
  - Wrap each individual specimen in pre-cleaned aluminum foil and freeze the sample as soon as practical
  - Take care to avoid cross contamination during sampling and handling
  - Clean sampling equipment, such as shovels, between collections
  - Ship bivalve samples on ice overnight, if possible to the laboratory conducting the analyses
- If increased sampling effort of tidal flats is deemed appropriate, follow sampling protocols as described earlier, but increase the number of quadrats and cores collected within intertidal flat transects. Mark the start of the transect and walk on a straight-line perpendicular to the shore to the farthest end of the tidal flat. Starting from the end, take samples from undisturbed areas as described above at specific intervals (e.g., 10 m) within the transect. At each sampling point, record the approximate distance from the shore transect marker, and record GPS coordinates. Tidal flat sampling should be done during the lowest tide possible.
- If possible, store samples from unoiled areas in one set of coolers, with oiled samples in a separate set of coolers.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each intertidal sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (infauna)
  - Sample #, date/time
  - Sampling method (core, excavation), sample collection depth, and core size (m^2)
  - Sediment oiling conditions (using standard shoreline assessment terminology), tidal elevation, weather conditions (e.g., wind direction and speed), presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, vertical changes in sediment characteristics, etc.

- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution in sand beach and tidal flat areas is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oiled samples separate from lightly or unoiled samples to reduce risk of cross-contamination.
- Immediately place all infauna samples in cooler and keep at 4°C. DO NOT FREEZE samples preserved in formalin. Tissue samples for chemical analysis can be frozen.
- Freeze samples for chemical analysis as soon as practical or by the end of each day if samples are not going to be analyzed within 7 days of collection.
- Tape lids on sample bottles so that they do not accidentally come off.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical, overnight (preferred), with completed Chain of Custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed, and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship any samples preserved in formalin or other chemicals as hazardous goods.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Analytical Methods

- Refer to those under Shellfish Tissue, Subtidal, and Intertidal Sediment guidelines.

Key References

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Appendix A
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil, or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:

- Beach Survey Field Data Form – 3 parts: Survey set-up, Surface quadrat, subsurface quadrat or core

Beach/Tidal Flat Survey Field Data Sheet - Set-up (one per transect)

1 Date: ____ Time: _____ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

4 Transect ID: _____

Total Transect Length (m): _____

5 Front Stake Way Point # _____ Latitude (DD) _____ Longitude (DD) _____

Back Stake Way Point # _____ Latitude (DD) _____ Longitude (DD) _____

6 Distance from shoreline to the midpoint of plots in each zone (m):

Supratidal _____ Middle Intertidal _____

Upper Intertidal _____ Low Intertidal _____

7 Photos

Front stake looking inland: _____ Back stake looking shoreward: _____

Front stake looking right: _____ Front stake looking left: _____

Offshore looking inland: _____

8 Plot-wide Sediment and Topography

Primary sediment type: _____ Secondary sediment type: _____

Presence of erosional scarps: yes / no Maximum vertical relief (cm) _____

9 Oiling Impact Extent

Sediment surface oiling coverage (%): _____

Surface oil thickness (circle one): Pooled Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

10 Transect sketch

Bearing to inland stake:

Drawing Legend	
FS	Front Stake
BS	Back Stake
----------	Transect
~~	Water line
□	Surface Quadrat
	Subsurface
■	Quadrat
○	Core Sample
X	Other sample

Notes including but not limited to presence, condition, and/or altered behavior of visible biota such as amphipods, bivalves, etc.

Beach/Tidal Flat Survey Field Data Sheet - Surface Quadrat (one per quadrat)

1 Date: _____ Time: _____ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

4 Transect ID: _____ Quadrat ID: _____

Quadrat location: Supratidal/ High/ Middle/ Low Intertidal/ Tidal Flat

Quadrat Size (m^2): _____

Quadrat Photo: _____

Direction from transect: Left / Right (looking from water towards shoreline)

Distance from Transect: _____

5 Sediment type

Primary _____ Secondary: _____

6 Oiling Impact Extent

Sediment surface oiling coverage (%): _____

Surface oil thickness (circle one): Pooled Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

7 Quadrat Fauna Information

Photo Only Yes / No Photos:

Species Name	Number	Species Name	Number	Additional Information:

Notes:

Beach/Tidal Flat Survey Field Data Sheet - Subsurface Quadrat or Core (one per sample)

1 Date: _____ Time: _____ Site ID: ____ Team ID:

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

_____

4 Transect ID: _____ Quadrat/Core ID: _____

Sample Type (Circle One): Subsurface quadrat Core

Quadrat location: Supratidal/ High/ Middle/ Low Intertidal/ Tidal Flat Quadrat Size (m^2): _____

Quadrat Photo: _____

Direction from transect: Left / Right Distance from Transect: _____

(looking from water towards shoreline) _____

Sample depth (cm): _____

5 Sediment type

Primary _____ Secondary _____

6 Oiling Impact Extent

Sediment surface oiling coverage (%): _____

Subsurface oil evident: yes / no Describe:

7 Subsurface sample

Sample retained: yes / no Sample No.: _____

Sieved: yes / no Sieve screen size (mm): _____

Sample preserved: yes / no Preservation type: Formalin Other

Species	Size Units:	Weight Units:	Count	Additional Information:

Notes:

Guidelines for Collecting Ephemeral Data in the Arctic: GRAVEL BEACH INTERTIDAL COMMUNITIES

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guidelines Objectives

The primary objective of this document is to provide guidelines on collecting samples from gravel beaches during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Most beaches in this category are composed of mixed sand and gravel along the outer shore, with the gravel component mostly pebbles and cobbles in size. Intertidal gravel beach habitats in the high Arctic generally do not support resident faunal organisms because they are subject to high wave energy, freezing temperatures and ice scour. Gravel beaches south of the Bering Strait and gravel beaches composed of larger cobble may have associated communities of intertidal organisms. Methods for collecting oil and gravel sediments for chemical analysis are described in the Stranded Oil and Intertidal Sediment guidelines respectively.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in gravel beach intertidal habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and degree of oiling in gravel beach habitats
- Support oil environmental transport modeling by documenting where oil stranded onshore

Study exposure

- Document the presence/absence and species composition, and estimate the abundance or density of the gravel beach intertidal community
- Document the extent and duration of exposure to the spilled material
- Measure oil related compounds in biological tissues
- Support exposure modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical and biological characterizations

Collaboration

- Support other assessment efforts (see Intertidal Sediment and Shellfish Tissue guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Gravel beach intertidal communities are difficult to sample because of the inherent heterogeneity of oil distribution over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an gravel beach or lagoon
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Consult ESI maps, state wildlife guides and other resources to determine what fauna may be present in the sampling area. Adjust the sampling strategy to target key species of interest, if present.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of gravel beach contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per location needed to respond to the sampling objectives.
- A stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected, is recommended if no other sampling strategy has been developed. This type of sampling improves the representative quality of samples by reducing sampling error (variability).
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling transects, transect spacing, sampling zone width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with gravel beach infauna sampling. Tarballs, sheens, or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.

- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars and aluminum foil for sample storage, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what locations have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled gravel beaches that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiling samples from sensitive/productive areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Gravel beach intertidal community samples should be collected pre-oiling, if possible, as soon as possible after oiling and periodically thereafter. Sampling frequency is a function of oil persistence,

biological community, habitat importance, and resource availability and should be defined in the study design.

- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for sampling gravel beach intertidal communities is at least three samples per tidal zone per location of relatively uniform oiling exposure. If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiling location and two heavily oiled locations.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.
- Oiled locations should be selected to represent different degrees of oiling (Heavy, Moderate, Light) that have similar degrees of exposure to wave energy because in gravel beaches oil persistence and the intertidal communities can vary widely in exposed versus sheltered settings. When choosing locations, consider that oiling may not be visible, especially on beaches with larger gravel or cobble.

Collaboration

- Gravel beach intertidal community samples can be collected in conjunction with intertidal and subtidal sediment sampling, nearshore water sampling, and stranded oil sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts are important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for infauna sampling under extreme cold conditions
- Plastic bags
- Surveying supplies – site markers (appropriate for substrate type), surveying flags and tape, quadrats (0.5 m^2 and 1.0 m^2), 30 m fiberglass tape measure marked in cm, hand counter
- Pencils, waterproof pens, waterproof labels, markers
- Large tub or bucket
- Pre-cleaned aluminum foil
- Sample bags/jars
- Shovel
- Hand coring device
- Sieve with 0.5 mm screen
- Tripod or quadrapod
- 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal) – preservation; and a stain (such as Rose Bengal) if appropriate
- Field Sample forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- GPS, camera (with spare batteries), and photo scales
- Shoreline terminology code list and shoreline assessment survey guidelines (see Stranded Oil guideline)

- Field sample forms (gravel beach fauna identification field guides/charts), field notebook (waterproof paper), shoreline oil terminology code list, other guidelines as needed (Field Photography, Subtidal and Intertidal Sediment guidelines)
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.
- The only equipment to be used between sites are a shovel and a hand corer, which should be cleaned with soap and clean water. Repeated digging in clean sediments can be a last resort for cleaning the shovel if soap or clean water are not available. Alternatively, use a clean dry towel or other dry material to clean the shovel before its next use. Additional cleaning may be required when working at oiled sites (see below).
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on boots, shovels and other equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Study Design Implementation

- If recent SCAT data are available, it may not be necessary to conduct and overflight or ground survey. Otherwise, if practical, conduct an overflight of the entire affected area within two hours of low tide (before or after) to locate gravel beaches and observe the extent of visible contamination. Note that weather, particularly fog, may dictate survey times that may not coincide with tides. Tides may not be a consideration along high Arctic shorelines where tidal range is small and water level is primarily wind driven.

- If an aerial survey is not feasible, survey from the ground. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs can be geo-referenced later. Observations should include:
  - Locations and approximate lengths of gravel beaches in the oiled area
  - Approximate degree of oiling of these habitats (following standard SCAT terminology; see Stranded Oil guideline)
  - Locations of access points, major landmarks, and potential ground-truth and reference stations
- Consider the accessibility of the locations and methods of access (e.g., plane, helicopter, boat, foot, etc.), and take all necessary precautions to ensure that access can be achieved safety.
- At each selected sampling location, conduct a preliminary visual survey of the shoreline and draw a field sketch showing:
  - Shoreline orientation, gravel size, supratidal washovers, etc. (see photographs)
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)

- At each selected site, establish at least three transects at a minimum of 30 m spacing apart.
- If no other sampling strategy has been developed, use a stratified random sampling approach by randomly selecting the first transect starting point and expanding the sampling site (other transects) systematically into a grid from that initial point. Randomly select sampling sites within each intertidal zone intervals.

- When establishing ground transects:
  - Record transect location using a GPS, but also accurately plot the transect on a map or aerial photograph (if available)
  - Permanently mark transect locations using “front and back” stakes that line up along the transect lines, and consider stake placement carefully to minimize loss to vandalism, erosion, ice-scouring, etc. Label the stakes to clearly identify the site as a NRDA sampling location. Assure that stakes do not present a hazard to people traveling by ATV or snow machine. On high-energy gravel shorelines, it may only be feasible to place one stake in the supratidal zone
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one stake is lost; note whether the angle reading is magnetic or corrected to true north
  - Take photographs of the transect at the beginning, middle and end, including upslope, downslope, and alongshore images. This takes little time and establishes a reference for future work
- If possible, run transects so that the lower intertidal area can be sampled within two hours of low tide (before or after). In the Chukchi and Beaufort seas the tide range is small (<30 cm); wind-driven storm surge is the most determinant factor in water height and should be considered for sampling. Tides are only a consideration south of the Bering Strait where the tidal range is greater than 1 m.

- Transects should be run perpendicular to the shoreline and encompassing the entire intertidal zone, including the supratidal zone (Figure 1). This is important because in the Arctic storms are likely to push oil into the suptratidal zone.

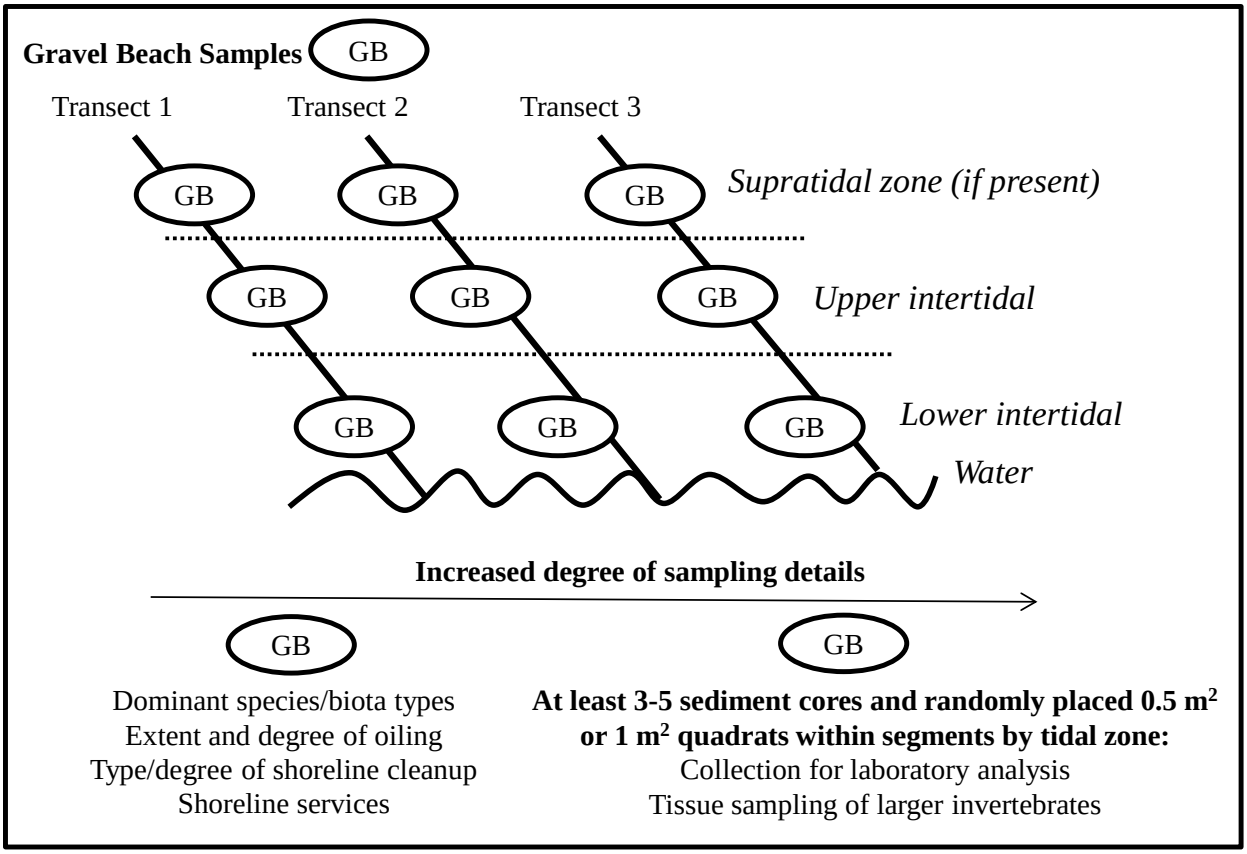

Figure 1. Schematic representation of the recommended gravel beach sampling strategy, including transects and sampling sites. Dashed lines represent approximate tidal zones. GB= gravel beach. Area-specific modifications may be needed. For example at locations with very narrow intertidal zones, only one or two sampling intervals may be used.

- On some high-Arctic shorelines, the intertidal zone may be very narrow. If this is the case, two sampling intervals can be defined on each transect, in the upper (or storm surge) and lower intertidal zone, or just one station if the intertidal zone is very narrow.
- Along each transect, use standard oiling terminology codes and estimation charts to record in field sample forms or field notebooks:
  - Date, time, weather conditions (e.g., wind direction and speed), tide level (as observed), and initials of observers
  - Distance of the interval
  - Physical setting (shoreline orientation, exposure to wave energy and tidal currents, etc.)
  - Length of the transect (in meters) and of the sampling zone
  - Sediment type and gravel average size (diameter in cm)
  - Dominant species or types of biota present (including signs of gravel intertidal organisms
  - Presence, condition, and/or altered behavior of visible biota such as amphipods, gastropods, crabs, etc.

  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts; see Stranded Oil guideline) and depth of oil penetration into the sediments, if any (using a shovel or coring device)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Presence of snow or ice along shoreline
  - Presence of biological resources and other relevant information
- Collect samples from each intertidal zone (described below) and record the distance along the transect and GPS coordinates of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If time allows, a calibration exercise is recommended prior to field sampling to ensure that all field teams consistently perform gravel intertidal community sampling.
- If sampling sites are covered by snow or ice, carefully remove the snow/ice without mixing or disturbing the sediment underneath and proceed with infauna sampling. Note snow and ice conditions on the field data sheet. If snow or ice in the intertidal zone are impacted by oil (as opposed to deposited on top of oiled sediments), it may be desirable to collect snow/ice samples for chemical analysis (see the Snow and Ice guidelines).
- If plant or animal life is expected to occur, or is found during data collection, sampling can be conducted to document the presence, composition, and general abundance of organisms.
- If highly quantitative density estimates are needed, or detailed comparisons of oiled vs. un-oiled locations are planned, an experienced gravel beach ecologist and statistician should be consulted to plan more detailed studies. If one is not available, the following sampling should be performed.
- For semi-quantitative surveys of selected shoreline transects or habitat locations:
  - Record the distance along the transect and GPS coordinates of each collection site
  - For species presence/absence, composition, or density estimates use quadrats (0.5 m^2 or 1.0 m^2). Randomly place the quadrat 1 and 2 m to the left and right of each transect by tidal zone (high, middle, low), for a n=4 per tidal elevation. Quadrats should be placed within 5 m of the transect line. One approach to random sampling is as follows:
    - Place two tape measures at a 90°angle along two sides of the sampling location
    - Use a random numbers table to randomly select two numbers
    - Place the center of the quadrat at the intersection of the two numbers on the tape measure
  - Photo-document the sampling locations, sample collection methods, organisms observed, any obvious oil impacts, etc. and keep a good record of the quadrat-photo sequence
  - Take high-resolution vertical photographs of each quadrat, if possible using a tripod or quadrapod, and record GPS coordinates. This will facilitate computer-based analyses of individual quadrats. When taking photographs observe the following:
    - If the presence of large or abundant motile invertebrates block the view of sessile invertebrates, carefully remove these organisms from the quadrats prior to taking photographs

and without disturbing less motile invertebrates, but make sure to document the removal of organisms in the field sample form
    - High-resolution photographs must include all four sides of the quadrat as these will be used to digitally count individuals and measure their coverage on a computer screen
    - When photographing highly dense quadrats, quadrat frames can be split into 2-sided frames to facilitate computer-based analyses
    - Photographs need to be relatively flat so that the entire quadrat falls within a similar focal plane, with minimal shadowing from crevices or projections. Photographs should be directly perpendicular to the quadrat
    - Ideally, photographs should be taken during the lowest tide and best light conditions (e.g., closest to midday or when overcast). Avoid shooting into the sun and avoid including sky, ocean, or tidepools in the view
    - If possible, use a quadrapod apparatus to support the camera at a constant height (1 m with a 35 mm lens) from the quadrat and position it to capture all four corners of the quadrat:
        - A quadrapod consists of a gray PVC or gray Schedule 80 PVC pipe frame with a photoplot-size bottom (0.5 m^2 or 1.0 m^2 internal dimensions) connected by 4 poles to the frame supporting the digital camera
        - Strobes mounted laterally and away from the camera can enhance lighting on the quadrats and reduce shadows
    - The best quality photographs are obtained by optimizing the ISO, aperture, and shutter speed
    - Remember that all quadrat images must be of sufficient quality to allow a positive identification and enumeration of the species in the quadrats
  - Estimate the percent cover of sessile species and algae within each quadrat
  - If time allows, identify (to the lowest taxonomic level practical), count and/or measure the size of mobile organisms (crabs and snails) captured either on the surface or under the rocks, and release them live; alternatively, place samples in plastic bags or jars and freeze, or preserve for identification in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate
  - Photo-document the sampling locations, sample collection methods, organisms observed, any obvious oil impacts, etc. and keep a good record of the quadrat-photo sequence
- If the gravel beach is conducive to coring (e.g., locations with mixed sand/gravel or where gravel over finer sediments can be easily moved to the side), or if requested in the sampling plan, use a hand coring device, typically a cylinder with an open cross-sectional area of 0.01 m^2 and a small hole on the top, to sample macroinfauna. A special corer type (clam corer) may be required to sample gravel beaches:
  - Prior to collecting cores, carefully remove large gravel, debris, shells, and other material without disturbing the sediments
  - Randomly place the corer 1 and 2 m to the left and right of each transect by tidal zone for a n=4 per tidal elevation sampled (note that all tidal elevations may not be sampled in this manner) Random selection of the sampling site can be performed following the guideline provided above
  - Take photographs and record the GPS coordinates of each sediment core site
  - Insert the corer into the sediment (in a vertical position) to a depth of 25 cm. Mark the outside of the corer so the depth of the core sample can be recorded, and so that cores do not exceed 25 cm in depth
  - Place a finger or thumb over the small hole (or insert a rubber stopper) to create a vacuum and extract the core
  - Empty the contents of the core into a labeled container, such as a large plastic bag
  - Describe the sediment composition and grain size, and sediment oiling, including depth of oil penetration

  - If the sediment size allows oil to penetrate deeper than the core depth, use a shovel to dig until the barrier layer (sand, mud or bedrock) is reached and describe oil penetration depth (see Stranded Oil guideline)
  - Core samples can be initially sieved in the field (using a 0.5 mm screen). When sieving, gently force water up through the bottom of the sieve, by bobbing the sieve up and down in a large bucket or tub of water, this prevents forcing animals into or through the bottom of the sieve
  - After sieving, place samples in a sample bag or jar and preserve with 10% buffered formalin
  - If field sieving of cores is not possible, preserved whole in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate
  - Place a waterproof label with the station location, sample number, and date inside the sample container
- To sample larger macroinfaunal target species, such as bivalves, if any, from gravel beaches conducive to this type of sampling:
  - Randomly place the quadrat 1 and 2 m to the left and right of each transect by tidal zone (high, middle, low), for n=4 per tidal elevation. Random selection of the sampling site can be performed following the guideline provided above
  - Take high resolution photographs of each quadrat, if possible using a tripod, and record GPS coordinates. High resolution photographs must include all four sides of the quadrat as these may be used to digitally count individuals and measure their coverage on a computer screen. Quadrat frames can be split into 2-sided frames to facilitate computer-based analyses. The best quality photographs are obtained by optimizing the ISO, aperture and shutter speed. Follow general guideline provided above
  - Record the quadrat GPS coordinates
  - Prior to removing quadrat contents, carefully remove large gravel, debris, shells, and other material without disturbing the sediments
  - Use a shovel to excavate the sediments within the quadrat to the appropriate depth, commonly 20-30 cm
  - If practical, sieve the excavated sediments using a 5 mm screen (or larger, depending on the size range of the target species)
  - Alternatively, spread out the excavated sediments and carefully pick all target species in the sample
  - Identify, count, weigh, and/or size the captured organisms in the field, and release them live, or, place samples in plastic bags or jars and freeze, or preserve in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal), and stain (such as Rose Bengal) if appropriate
  - Place a waterproof label with the station location, sample number, and date inside the sample container
- If tissue analyses are planned for organisms, such as mussels, follow the Shellfish Tissue guideline. Tissue samples may be collected particularly from areas with obvious oiling impacts. Briefly:
  - Wrap each individual specimen in pre-cleaned aluminum foil, and freeze the sample as soon as practical
  - Take care to avoid cross contamination during sampling and handling
  - Clean sampling equipment, such as shovels, between collections
  - Ship biological samples on ice overnight, if possible to the laboratory conducting the analyses
- If collection of gravel for chemical analyses is required by the sampling plan, avoid placing samples in large glass jars because these may break during sampling, transport, or storage. Alternatively, use the smallest possible glass jars, and pack them with enough small gravel to fill the entire jar without leaving room for gravel movement. Cap the jars and carefully wrap them individually in bubble wrap or other protective material, and store them in a box with cardboard dividers leaving every other

space empty. Alternatively, use small Teflon bottles (less ideal). For larger gravel size, double wrapped gravel in clean aluminum foil and put in a bag (see Intertidal Sediment guideline).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each intertidal sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (fauna)
  - Sample #, date/time
  - Sampling method (core), sample collection depth, and core size (m^2)
  - Sediment oiling conditions (using standard shoreline assessment terminology), tidal elevation, weather conditions (e.g., wind direction and speed), presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
  - Sediment characteristics: gravel size, texture, color, biota, vegetation, debris, odor, etc.
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution in gravel beaches is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oiled samples separate from lightly or unoiled samples to reduce risk of cross-contamination.
- Samples should be received by the laboratory for processing within 7 days of collection.
- Immediately place all invertebrate samples in cooler and keep at 4°C. DO NOT FREEZE samples preserved in formalin. Tissue samples for chemical analysis can be frozen.
- Ship any samples preserved in formalin or other chemicals as hazardous goods.
- Tape lids on sample bottles so that they do not accidentally come off.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical, overnight (preferred), with completed Chain of Custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect

them from breakage, shipping containers are sealed, and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.

- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Analytical Methods

- Refer to those under Shellfish Tissue and Intertidal Sediment guidelines.

Key References

Eleftheriou, A. and N.A. Holme. 1984. Macrofauna techniques. Chapter 6. In: N.A. Holme and A.D. McIntyre (eds.), Methods for the Study of Marine Benthos, IBP Handbook 16, Blackwell Scientific Publications, Oxford, UK. pp 140-216.

NOAA Damage Assessment Center. 1997. Field forms and codes. Appendix 6, In: Natural Resource Damage Assessment Emergency Guidance Manual, Version 3.1. NOAA Damage Assessment Center, Silver Spring, MD.

Wolff, W.J. 1987. Flora and macrofauna of intertidal sediments. Chapter 4. In: J.M. Baker and W.J. Wolff (eds.), Biological Surveys of Estuaries and Coasts, Estuarine and Brackish Water Sciences Association Handbook, University of Cambridge Press, Cambridge, UK. pp. 81-105.

Appendix A
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil, biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:

- Beach Survey Field Data Form – 3 parts: Survey set-up, Surface quadrat, subsurface quadrat or core

Gravel Beach Survey Field Data Form - Set-up (one per transect)

1 Date: ___ Time: ___ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

4 Transect ID: _____

Total Transect Length (m): _____

5 Front Stake Way Point # _____ Latitude (DD) _____ Longitude (DD) _____

Back Stake Way Point # _____ Latitude (DD) _____ Longitude (DD) _____

6 Distance from shoreline to the midpoint of plots in each zone (m):

Supratidal _____ Middle Intertidal _____

Upper Intertidal _____ Low Intertidal _____

7 **Photos**

Front stake looking inland: _____ Back stake looking shoreward: _____

Back stake looking right: _____ Front stake looking left: _____

Offshore looking inland: _____

8 **Plotwide Sediment and Topography**

Primary sediment type: _____ Secondary sediment type: _____

Presence of erosional scarps: yes / no Max. vertical relief (cm) _____

9 **Oiling Impact Extent**

Sediment surface oiling coverage (%): _____

Surface oil thickness (circle one): Pooled Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

10 **Transect Sketch**

Bearing to inland stake: _____

Drawing Legend	
FS	Front Stake
BS	Back Stake
----------	Transect
~~	Water line
□	Surface Quadrat
■	Subsurface Quadrat
○	Core Sample
X	Other sample

Notes: including but not limited to presence, condition, and/or altered behavior of visible biota such as bivalves, etc.

Gravel Beach Survey Field Data Form - Surface Quadrat (one per quadrat)

1 Date: ___ Time: ___ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

_____

4 Transect ID: _____ Quadrat ID: _____

Quadrat location: Supratidal/High /Middle /Low Intertidal Quadrat size (m^2): _____

Quadrat photo: _____

Direction from transect: Left / Right Distance from transect: _____

(looking from water towards shoreline)

5 Sediment Type

Primary: _____ Secondary: _____

6 Oiling Impact Extent

Sediment surface oiling coverage (%): _____

Surface oil thickness (circle one): Pooled Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

7 Quadrant Fauna Information

Photo Only Yes / No Photos:

Species Name	Number	Species Name	Number	Additional Information:

Notes:

Gravel Beach Survey Field Data Form - Subsurface Quadrat or Core (one per sample)

1 Date: ___ Time: _____ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

_____

4 Transect ID: _____ Quadrat/Core ID: _____

Sample type (Circle One): Subsurface quadrat Core

Quadrat location: Supratidal/ High /Middle /Low Intertidal Quadrat size (m^2): _____

Quadrat photo: _____

Direction from transect: Left / Right Distance from transect:

(looking from water towards shoreline)

Sample depth (cm): _____

5 **Sediment type**

Primary: _____ Secondary: _____

6 **Oiling Impact Extent**

Sediment surface oiling coverage (%): _____

Subsurface oil evident: yes / no

7 **Subsurface sample**

Sample retained: yes / no Sample No.: _____

Sieved: yes / no Sieve screen size (mm): _____

Sample preserved: yes / no Preservation type: Formalin Frozen Other

Species	Size Units:	Weight Units:	Sex	Count	Additional Information:

Notes:

Guidelines for Collecting Ephemeral Data in the Arctic: ROCKY INTERTIDAL HABITATS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting rocky intertidal community samples from intertidal or shallow subtidal areas during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Arctic rocky shores are very different from rocky shores in other regions. There is very little rocky shore north of the Bering Strait. The predominant areas where there are rocky shores in the Bering Strait and south of the Bering Strait are islands (Saint Lawrence Island, Little Diomede Island, Fairway Rock, Sledge Island, and King Island). With the exception of Saint Lawrence Island, the coasts of these islands are very steep and do not have flat benches. Many rocky areas are made up of large rocks from the size of cobbles to boulders or steep rocky shores. Rocky shorelines are also present along the coast of the Bering Sea, including in Norton Sound, Bristol Bay and the Aleutian Islands. Other related habitats (e.g., gravel beaches) are covered in separate guideline documents.

Sampling Objectives

Characterize oil

- Determine the composition of oil compounds in rocky intertidal habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and degree of oiling in rocky intertidal habitats
- Document the presence/absence and species composition, and estimate the abundance or density of the invertebrate and algal community associated with rocky intertidal habitats
- Support oil environmental transport modeling by documenting where oil stranded onshore

Study exposure

- Support exposure and transport modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical and biological characterizations

Collaboration

- Support other ongoing ephemeral sampling efforts (see Intertidal and Subtidal Sediment guidelines)
- Collect tissue samples for chemical analysis (see Shellfish Tissue guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.
- Sampling rocky intertidal habitats by boat is not recommended unless the area is sheltered and the water very calm.
- Special precautions are to be taken when sampling rocky intertidal habitats as sampling areas may have steep rocky shores. In areas relatively free of ice scour, the exposed rocky areas will be covered by macro-algae which will make sampling challenging. Modifications to these guideline documents will be needed to adjust for sampling in these areas.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Rocky intertidal habitats are difficult to sample because of the inherent heterogeneity, steep slopes, and in some cases difficult access.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as intertidal boulders or cobble beaches
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of rocky intertidal habitat contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per site needed to respond to the sampling objectives.
- A stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected, is recommended if no other sampling strategy has been developed. This type of sampling improves the representative quality of samples by reducing sampling error (variability).
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling transects, transect spacing, intertidal zone width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field. This is particularly the case for rocky intertidal habitats, as sampling strategies may need to be modified to sample each type of rocky shore (steep rocky shores, cobble/boulder rocky shores, irregular rocky shores with crevices, etc.).

- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with rocky intertidal sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels; solvent rinsing of jars, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Rocky intertidal habitats can be: 1) rocky shores composed of steep rock walls and vertical bedrock exposed to moderate to high wave energy (see photograph); 2) platforms consisting of wave-cut or low-lying bedrock exposed to moderate to high wave energy; and 3) rocky shores characterized by vertical rock walls, bedrock outcrops, wide rock platforms and boulders found in sheltered areas and generally protected from wave

exposure.

- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data or other tools to determine what location have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, shipping, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled rocky intertidal areas. Collecting pre-oiled rocky intertidal samples from areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled "control" areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Steep rock walls that are exposed to wave action and ice scouring will not retain oil and will generally have limited biological productivity. These rocky shores are likely to be challenging to access. Consider prioritizing sampling in other rocky intertidal habitats (cobble, boulder, bedrock platforms or protected rocky habitats), or in other habitat types before attempting to sample steep, exposed rock walls.
- Rocky intertidal samples should be collected pre-oiling, if possible, as soon as practical after oiling and periodically thereafter. Sampling frequency is a function of oil persistence, biological community composition, habitat importance, and resource availability and should be defined in the study design.
- Oiled locations should be selected to represent different degrees of oiling (Heavy, Moderate, Light) that have similar degrees of exposure to ice scour and wave energy because the rocky intertidal communities can vary widely in exposed versus sheltered settings. When sampling oiled areas focus first on the sheltered rocky shores as that is where most of the oil will likely end up for the longest period of time. A pre-survey or detailed walk of the area may be needed to select these areas appropriately. Information about exposure and oil retention potential is also available for many shorelines in ShoreZone.
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting rocky intertidal samples is at least three samples per intertidal zone per location of relatively uniform oiling exposure. If relevant data are available, a power analysis or other modeling approaches should be used to determine the number of samples needed before going into the field.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration

- Rocky intertidal samples can be collected in conjunction with intertidal sediment (if any), stranded oil, and gravel beach sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs– for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)

- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for sampling under extreme cold conditions
- 8 oz certified organic-clean jars with Teflon-lined lids and labels – for oil samples
- Surveying supplies – site markers (appropriate for substrate type), surveying flags and tape, quadrats (0.5 m^2 and 1.0 m^2), portable hammer drill with drill bits, stainless steel bolts, marine epoxy (or similar), 30 m fiberglass tape measure marked in cm, hand counter, caliper
- Pencils, waterproof pens, waterproof labels, markers
- Evidence tape (see Chain of Custody guidelines)
- Small containers– for holding motile invertebrates
- Aluminum foil: the dull side should be pre-cleaned with acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) to wrap samples of larger gravel, with the dull side in contact with the sample
- Ziploc or Whirl-Pak bags, and additional sampling jars
- 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal)– preservation; and a stain (such as Rose Bengal) if appropriate
- Field Sample forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Rocky intertidal identification field guides/charts (if available), field notebook (waterproof paper), other guidelines as needed (Field Photography, Subtidal and Intertidal Sediment, Stranded Oil guidelines)
- GPS, camera (with spare batteries), and photo scales
- Single or double strobe lighting, quadrapod frame for photoplots
- Tape measure and ruler
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Study Design Implementation

- Ice scouring and freezing temperatures affect the distribution and abundance of rocky intertidal organisms in the Arctic. Ice scour on flat surfaces likely occurs on a yearly basis in many rocky intertidal locations and, consequently, the biota likely to occur on these rocky shore types are either

ephemeral (e.g., diatomaceous scum) or mobile (e.g., gastropods/crustaceans). Sessile invertebrates and organisms less tolerant to direct cold air exposure are likely to be found in crevices and other areas sheltered from ice scour. Consequently, sampling crevices is an important component of rocky intertidal sampling (see below).

- If recent SCAT data are available, it may not be necessary to conduct and overflight or ground survey. Otherwise, if practical, conduct an overflight of the entire affected area within two hours of low tide (before or after) if possible, to observe the extent of visible contamination. Note that weather, particularly fog, may dictate survey times that may not coincide with tides. Tides may not be a consideration along high Arctic shorelines where tidal range is small and water level is primarily wind driven.
- If an aerial survey is not feasible, survey from the ground. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs can be geo-referenced later. The use of high definition video is also recommended. If an overflight is not feasible, conduct a pre-walk of the area. Observations should include:
  - Locations and approximate lengths of rocky intertidal habitats in the oiled area
  - Approximate degree of visible oiling of these habitats (if possible)
  - Locations of access points, major landmarks, and potential ground-truth and reference locations (if possible)

Note: Overflights may not be adequate enough to assess characterize rocky intertidal habitat oiling. It is highly advisable to consult with local experts and to select sampling sites based on trajectory modeling or SCAT data.

- Upon return, select locations for ground surveys based on degree of oiling (Heavy, Moderate, Light, No Oil; standard SCAT terminology), degree of rocky shoreline exposure and physical characteristics (e.g., steep shores, cobble/boulder rocky shores).
- If no other sampling strategy has been developed, use a stratified random sampling approach by randomly selecting the first transect starting point and expanding the sampling site (other transects) systematically into a grid from that initial point. Randomly select sampling sites within each intertidal zone intervals.
- Based on the study design and/or sampling strategy outlined before going into the field, establish a minimum of three transects spaced at least 30 m apart (recommended). Transects should be perpendicular to the shoreline and encompass the entire intertidal zone.
  - Record transect location using a GPS, but also accurately plot the transect on a map or aerial photograph
  - Permanently mark transect locations using “front and back” stakes which line up along the transect lines, and consider stake placement carefully to minimize loss to vandalism, erosion, ice-scouring, etc. Label the stakes to clearly identify the site as a NRDA sampling location to avoid possible confusion with the multitude of other survey stakes found across the Arctic
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one stake is lost; note whether the angle reading is magnetic or corrected to true north
  - Take photographs of the transect at the beginning, middle and end, including upslope, downslope, and longshore images. This takes little time and establishes a reference for future work
- Divide each transects into sampling intervals based on the intertidal zones: upper intertidal, middle intertidal and lower intertidal.
- On some high-Arctic shorelines, the rocky intertidal zone may be very narrow. If a rocky intertidal transect is too narrow to have three sampling intervals on, consider running the transect from the supratidal or storm surge line (usually demarcated by a line of logs or debris) to the lower intertidal. Alternately, two sampling intervals can be defined on each transect, in the upper (or storm surge) and lower intertidal zone, or just one station if the intertidal zone is very narrow.

- Ground-based ephemeral data collection on steep rocky cliffs may not be feasible or necessary; visual surveys from vessels or aircraft may be the preferred method for assessing this habitat. If ground-based surveys are conducted, some of the guidelines for studying intertidal rocky shorelines may not be applicable to steep cliffs or will need to be adapted for safety and feasibility. On steep rocky shorelines, transects will be vertical or near vertical and tidal zones will be defined by biota bands or height above the water. As with less steep shorelines in the high Arctic, the tidal zone may be too narrow to divide into multiple sections.
- When establishing ground transects:
  - Record transect location using a GPS, but also accurately plot the transect on a map or aerial photograph (if available)
  - Permanently mark transect locations using "front and back" stakes which line up along the transect lines, and consider stake placement carefully to minimize loss to vandalism, erosion, ice-scouring, etc. Label the stakes to clearly identify the site as a NRDA sampling location. If marking transects with stakes is not possible or unpractical, the following alternate approaches may be used:
    - Use a small amount of epoxy to mark rocks at the transect ends. This can be done by pressing a blob of well-mixed epoxy onto a clean rock surface (important to ensure adhesion) to form a mound approximately 4 cm in diameter
    - Use water resistant paint to mark rocks at the transect ends. Strong colors (red, orange) are preferred
    - Use a hand drill to drill small holes on the rock, and use epoxide to permanently put bolts in place. Consider their placement to make sure these markers are not a tripping hazard. Use the primary bolt marking the site from which other lines can be found. Note that bolts may not be appropriate in ice scouring areas
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one stake is lost; note whether the angle reading is magnetic or corrected to true north
  - Take photographs of the transect at the beginning, middle and end, including upslope, downslope, and longshore images. This takes little time and establishes a reference for future work
- For each transect, record:
  - Date, time, weather conditions (e.g., wind direction and speed), and tide level
  - Physical setting (shoreline orientation, exposure, etc.)
  - Substrate type and grain-size (if applicable) (e.g., mud, sand, granule, pebble, cobble, boulder)
  - Length of the transect (in meters) (if applicable) and of the sampling zone
  - Extent and degree of visible shoreline oiling (use SCAT guidelines for a more detailed assessment of shoreline oiling if needed)
  - Extent and degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Presence of snow or ice along shoreline
  - Presence of biological resources or other relevant information
  - Presence, condition, and/or altered behavior of visible biota such as amphipods, gastropods, crabs, etc.
- Collect samples from each rocky intertidal zone (described below) and record the distance along the transect and GPS coordinates of each sampling site.
- Take pictures of the transect before and after sampling and pictures of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.

- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If time allows, a calibration exercise is needed to ensure that all field teams consistently perform sampling.
- At each selected sampling site, conduct a preliminary visual survey of the shoreline and draw a field sketch showing:
  - Shoreline orientation, type, grain size, etc.
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts)
  - Type or degree of shoreline cleanup, if already performed (particularly note substrate disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Location of transects
- Sampling is conducted to document the presence, composition, and general abundance of organisms including barnacles (e.g., *Chthamalus* and *Balanus*), mussels, and serpulid worms, among others. If highly quantitative density estimates are needed, or detailed comparisons of oiled vs. un-oiled sites are planned, an experienced rocky intertidal ecologist and statistician should be consulted to plan more detailed studies. For surveys of selected shoreline transects or habitat areas:
  - Record the distance along the transect and GPS coordinates of each collection site
  - For species presence/absence, composition, or density estimates use quadrats (0.5 m^2 or 1.0 m^2). Randomly place the quadrat 1 and 2 m to the left and right of each transect by tidal zone (high, middle, low), for a n=4 per tidal elevation. If possible record a visual estimate of percent oiling by key species. ***Note***: A 2-dimensional quadrat frame may not be as effective when sampling a rocky shore with complex profiles, high scour rocky intertidal areas, and rock crevices. Under some situations, string quadrats may be more appropriate
  - Take high-resolution vertical photographs of each quadrat, if possible using a tripod or quadrapod, and record GPS coordinates. When taking photographs:
    - Take photographs of the quadrat to document motile and sessile organisms
    - When documenting coverage of sessile organisms, prior to taking photographs remove from the quadrats any large or abundant motile invertebrates blocking the view of sessile invertebrates and algae. Carefully remove these organisms without disturbing the sessile/less motile organisms. Make sure to document the removal of organisms in the field sample form from photoplots prior to photo/scoring
    - High-resolution photographs must include all four sides of the quadrat as these will be used to digitally count individuals and measure their coverage on a computer screen
    - When photographing highly dense quadrats, quadrat frames can be split into 2-sided frames to facilitate computer-based analyses
    - Photographs need to be relatively flat so that the entire quadrat falls within a similar focal plane, with minimal shadowing from crevices or projections. Photographs should be directly perpendicular to the quadrat
    - If possible, use a quadrapod apparatus to support the camera at a constant height (1 m with a 35 mm lens) from the quadrat, and positioned to capture all four corners of the quadrat:
      - A quadrapod, consists of a gray PVC or gray Schedule 80 PVC pipe frame with a photoplot-size bottom (0.5 m^2 or 1.0 m^2 internal dimensions) connected by 4 poles to the frame supporting the digital camera

        - Strobes mounted laterally and away from the camera can enhance lighting of the quadrats and reduce shadows
      - The best quality photographs are obtained by optimizing the ISO, aperture, and shutter speed. Most digital cameras take acceptable photographs when set to "auto", but make manual adjustments if needed
      - Remember that all quadrat images must be of sufficient quality to allow a positive identification and enumeration of the species in the quadrats
    - Estimate the percent cover of sessile species and algae within each quadrat. If sessile species are present in the form of layers, measure the thickness of the layer from the substrate to the top of the layer, and document the relative dimensions of the layers if these are present as distinct aggregations or patches
    - If time allows, identify (to the lowest taxonomic level practical), count and/or measure the size of mobile organisms (crabs and snails) captured either on the surface or under the rocks, and release them live; alternatively, place samples in plastic bags or jars and store in coolers. Preserve for identification in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal) as soon as practical, and stain (such as Rose Bengal) if appropriate
- Sessile invertebrates and organisms less tolerant to direct cold air exposure, ice scour and wave action are likely to be found in crevices, which depending on their configuration, have the potential to retain oil. Sampling crevices can be challenging and is best accomplished by counting the number of organisms or the percent cover of sessile organisms within fixed plots. Since this type of sampling may be obstructed in deep or narrow crevices, assessments are to be made based on what is visible to the sampler.
- At some locations where localized impacts are obvious, it may be necessary to sample intertidal pools, which are operationally defined as depressions with persistent water >5 cm deep. Sampling the biota of intertidal pools is challenging because of the three-dimensional nature and highly variable size of pools. When sampling intertidal pools:
  - Treat individual tidal pools as the sampling unit and assess the composition (abundance and percent cover; as describe above) of the entire community within each intertidal pool
  - If a intertidal pool is too large to sample the entire pool, randomly place quadrats within each pool (the number of quadrats will depend on the size of the pool; 1-3 quadrats, preferred), and collect quadrat information as described above
  - Take photographs of individual pools from different angles using a photo scale as a reference
- It is important to remember that sampling boulder rocky shores requires a modification of standard rocky intertidal sampling procedures because the three-dimension scales of these habitats make quantifying algae and sessile invertebrates challenging. One approach to sampling boulder rocky shores is to treat individual boulders as the sampling unit and assess the composition (abundance and percent cover; as describe above) of the entire community within each boulder. Within boulders of similar size, randomly select a minimum of 3 boulders to be sampled.
- If tissue analyses are planned for mollusks, follow the Shellfish Tissue guideline. Briefly:
  - Wrap each individual specimen in pre-cleaned aluminum foil, and freeze the sample as soon as practical
  - Take care to avoid cross contamination during sampling and handling
  - Clean sampling equipment, such as shovels, between collections
  - Ship biological and sediment samples on ice overnight to the laboratory conducting the analyses, or as soon as practical
- Take oil samples, particularly in areas where rocky intertidal impacts are visibly obvious (see Stranded Oil guideline).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.

- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each rocky intertidal sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (biota, tissue)
  - Sample #, date/time
  - Sampling method (quadrat)
  - Oiling conditions (using standard shoreline assessment terminology), tidal elevation, weather conditions, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
  - Grain size characteristics: grain size, texture
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution in rocky intertidal areas is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- Immediately place all samples in cooler and keep at 4°C. DO NOT FREEZE samples preserved in formalin. Tissue samples for chemical analysis can be frozen.
- Freeze samples for chemical analysis as soon as practical or by the end of each day if samples are not going to be analyzed within 7 days of collection.
- Tape lids on sample bottles so that they do not accidentally come off.
- If possible, store samples from unoiled locations in one set of coolers, with oiled samples in a separate set of coolers.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship any samples preserved in formalin or other chemicals as hazardous goods.

- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Analytical Methods

- Refer to those under Shellfish Tissue, Subtidal, and Intertidal Sediment guidelines

Key References

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Appendix A
Supporting Documentation – Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on water-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil, or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:

- Beach Survey Field Data Form – 2 parts: Survey set-up, Surface quadrat

Rocky Intertidal Habitat Survey Field Data Sheet – Set-up (one per transect)

1 Date: ___ Time: ____ Site ID: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

_____

4 Transect ID: _____

Total Transect Length (m): _____

5 Front Stake/Mark Way Pt. # ____ Latitude (DD) _____ Longitude (DD) _____

Back Stake/Mark Way Pt. # ____ Latitude (DD) _____ Longitude (DD) _____

6 Distance from high-tide line to the midpoint of plots in each zone (m):

Supratidal ____ Middle Intertidal _____

Upper Intertidal ____ Low Intertidal _____

7 Photos

Front stake/mark looking inland: ____ Back stake/mark looking shoreward: _____

Front stake/mark looking right: ____ Front stake/mark looking left: _____

Offshore looking inland: ____

8 Plot-wide Substrate and Topography

Primary substrate type: _____ Secondary substrate type: _____

Maximum vertical relief (cm) _____

9 Oiling Impact Extent

Substrate surface oiling coverage (%): _____

Surface oil thickness (circle one): Thick Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

10 Transect sketch

Bearing to back stake/mark: _____

Drawing Legend	
FS	Front Stake
BS	Back Stake
----------	Transect
~~	Water line
□	Surface Quadrat
■	Subsurface Quadrat
○	Core Sample
X	Other sample

Note presence, condition, and/or altered behavior of visible biota such as chitons, bivalves, crabs, etc.

Rocky Intertidal Habitat Survey Field Data Sheet – Surface Quadrat (one per quadrat)

1 Date: ____ Time: ____ Site ID: _____ Team ID:

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliations: _____

4 Transect ID: _____ Quadrat ID: _____

Quadrat Location: Supratidal/ High/ Middle/ Low Intertidal/ Quadrat Size (m^2): _____

Quadrat Photo: _____

Direction off transect (look landward): Left / Right Distance from Transect: ____

(looking from water towards shoreline)

5 Substrate type:

Primary: _____ Secondary: _____

6 Oiling Impact Extent

Substrate surface oiling coverage (%): _____

Surface oil thickness (circle one): Thick Cover Coat Stain N/A

Surface oil character (circle one): Fresh Mousse Surface residue Tar Asphalt N/A

7 Quadrat Flora Information

Photo Only Yes / No Photo file numbers: _____

Total vegetative cover (%): Total dead vegetative cover (%):

Species Name	Live cover (%)	Dead cover (%)	Additional Information:

8 Quadrat Fauna Information

Photo Only Yes / No Photos:

Species Name	Number, cover (%)	Species Name	Number, cover (%)	Additional Information:

Note presence, condition, and/or altered behavior of visible biota such as chitons, bivalves, crabs, etc.

Guidelines for Collecting Ephemeral Data in the Arctic: ICE

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collection of ice samples for chemical and biological analysis during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in the ice compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Characterize the physical characteristics of the ice habitat
- Support oil environmental transport modeling

Study exposure

- Document exposure of ice and under-ice organisms to oil compounds
- Support exposure modeling

Quality assurance/quality control

- Ensure the integrity the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical characterizations

Collaboration

- Support other ongoing efforts including, but not limited to, validation of remote sensing activities , modeling of impacts to water-column resources (see Water guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team. Special precautions are necessary for working on sea ice or in ice-infested waters.

- Sampling ice is a highly specialized activity that most field personnel are not familiar with. Plan to involve or consult with experts, such as the University of Alaska Sea Ice Group or others, before undertaking any ice sampling activities.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Ice samples are difficult to sample because of the inherent heterogeneity of oil distribution over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Information about the location and movement of oil under ice may be available from the emergency response effort. Oil is difficult to detect under ice and will tend to concentrate in certain areas depending on the under-ice roughness. Oil would be unlikely to spread evenly.
- Use a computer or conceptual model of the extent of ice contamination or an appropriate power analysis to estimate the number of locations and number of sites per location needed to respond to the sampling objectives.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Information from the national ice center, emergency response or other sources should be used to develop a sampling strategy and estimate distances and number of sampling locations before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with ice sampling. If observed during ice sampling, tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars for total hydrocarbons (THC) and polycyclic aromatic hydrocarbons (PAH) analyses, etc.

- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- Oil will not weather under ice at the rate that it does when exposed to air. Consider the stability of the oil contamination and ice-associated biological communities when planning ephemeral data collection in ice habitats. During freeze-up and break-up ice conditions may change very rapidly, which could influence sample timing.
- It is important to obtain reliable information and analytical chemistry data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- Ideally, ice cores should be collected separately for chemical and biological analysis. However, under some circumstances (including limited storage capacity; see below), the same ice core can be used for both chemical and biological analyses.
- If space is limited or time constraints exist, prioritize sample collection to obtain as much information as practical about resources at risk and their exposure to oil.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference locations.
- Use trajectory models, conceptual models, overflight information, remote sensing data or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled ice areas that are known to be biologically productive, or highly relevant for human use. Collecting pre-oiled ice from sensitive/productive areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled “control” areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.

- Ice samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting ice samples is at least three samples per location of relatively uniform oiling exposure.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration
- Ice samples can be collected in conjunction with water samples.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.
- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for water sampling under extreme cold conditions
- Sampling jars – certified organic-clean glass jars (solvent rinsed) with Teflon-lined lids and labels:
  - 1 L glass jars, amber glass preferred. ***Note:*** The mouth of the jar must be large enough to fit ice core without resourcing to core sectioning
- Shovel
- Ice drills including augers (mechanical or manual)/corers – for collecting ice cores
- Ice saw or knife– for ice core sectioning
- Teflon (PTFE) bags – 60 x 60 cm, organic clean (solvent rinsed) with closures or cable ties (preferred). Smaller sizes are also needed for ice-core segment storage
- Ice core storage boxes– for biological samples
- Ice probe for measurement of ice core temperature and salinity (preferred); a quick reading thermometer and a handheld salinometer (less ideal)
- 10 and 20 micron pore filters
- Glass or plastic vials- for sea-ice flora preservation
- Lugol, 10% buffered formalin (preferred), 95% ethanol (less ideal) – for sample preservation
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- Field notebook (waterproof paper)
- Pencils, waterproof pens, waterproof labels, markers
- Sorbent pads
- GPS, camera (with spare batteries), and photo scales
- Small drop camera with underwater capabilities (e.g., GoPro) – for under ice photography
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment is not available):

- Sufficient quantities of pre-cleaned or disposable single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Obtaining an adequate number of quality control samples is essential. At a minimum, a trip blank (accounts for contamination introduced during shipping and handling) and field blank (accounts for contamination introduced during sampling) should be maintained for each sampling effort and generally be collected at a rate of 5% and 10%, respectively, of all samples.
- Ideally, trip and field blanks are a sampling jar containing ultra-pure or distilled water. Blanks may be provided by the receiving laboratory.
- A trip blank is an unopened sampling jar and should be transported with the samples and remain sealed in the cooler during sampling activities.
- A field blank should be collected at approximately every third sampling site, or at least at an "un-oiled" and "oiled" site, by leaving the field blank sample jar open for the duration of the sampling period at that site. Record the site where field blanks were taken on the field sample form.
- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.
- Duplicate samples should be collected at every third sampling site or following the specifications of the work plan. A duplicate sample is collected from the same location and following the same steps as the preceding sample. This is not the same as collecting replicates from each site/depth. Duplicates should account for 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless specified in the work plan.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.

- The only types of equipment to be used between sites are ice drills/corers and saw or knife, which should be cleaned with soap and clean water. Alternatively, use a clean dry towel or other dry material to clean the equipment before its next use.
- Additional decontamination steps MUST be taken when using ice drills/corers and saw or knife when these become contaminated, and particularly when sampling in oiled sites. To decontaminate these tools prior to each use:
  - Wash sampling equipment with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used. If unfrozen water is not available, snow can also provide significant cleaning
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Potential sources of contamination while sampling from vessels or vehicles (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts, consider using non-motorized vehicles to access offshore ice sampling locations, and designate clean areas for sampling. Park aircraft, snow machines, boats or other vehicles at least 5 meters away and upwind from the sampling site. If possible, turn off vehicle engines while sampling.
- Take precautions to avoid cross-contamination of the site from oil on personal equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- When photographing areas or locations with sea ice consider including a photo scale, person or object in some pictures for perspective and scale.
- Collection of ice cores is a delicate process that requires previous coring experience or field calibration to ensure that the bottom of the ice core is collected as intact as practical. Note that full speed mechanical drilling disturbs the bottommost few centimeters more than hand drilling. Mechanical drilling may be preferred if the objective is to conduct sampling as quickly as practical, but the tradeoff may be loss of the most oil contaminated and biologically productive skeletal layer of ice.
- When collecting ice cores for chemical or biological analyses:
  - Ice cores can be collected using a wide spectrum of ice drills ranging from hand augers (useful when collecting cores from the top two meters of the ice sheet) to mechanical drills (useful when

collecting cores at greater depths within the ice sheet). Select the appropriate drill/coring device depending on area-specific characteristics and sampling objectives

- Avoid sampling ice cores under sheens and oil slicks, but if unavoidable, clear surface oil prior to sampling by sweeping the area with sorbents. If oil has reached the surface of the ice by moving through brine channels and it is not possible to clear the surface, it may still be important to collect a sample. Note this type of sample contamination in the field data sheet and in the field notebook
- Prior to coring, remove all surface snow and unconsolidated ice with a clean shovel
- Take photographs of the site before and after coring, as well as photographs of the core, and record the GPS coordinates of the core extraction site. In addition, take notes in the field notebook and take a picture of the field book before the core is extracted
- Care should be taken to prevent the contact of the outside of the core with the drilling equipment, drilling fluids, and other sources of contamination
- As soon as a core is taken, make sure that you process it as quickly as practical, as temperature and bulk salinity (brine drainage) will change on a short time scale
- If possible, it is recommended that one core be taken for temperature profiles and one for bulk salinity as recording the temperature can take a while and brine drainage can be significant in the bottom segments (see below)
- If possible, use a small drop camera (GoPro or other underwater camera) mounted on a pole to send down the core hole to take pictures of the surrounding ice to provide some indication of the representativeness of your sample. Biologically, the spatial scales of variability can be very small and influenced by the amount of snow overhead
- Record the external temperature at the time of core collection and extraction
- Following recovery, ice cores should remain in a frozen state until analysis, typically in a laboratory off the ice (preferred)
- Alternatively, ice cores can be melted and water stored for analysis (less ideal); however, all precautions have to be taken to avoid any sample contamination
- Do not expose any core collected for biological sampling to direct sunlight, as this can alter their biological makeup
- Collect at least three replicate sets of cores per location of homogenous oil exposure. Because even level sea ice is not homogenous at the ice-air interface, protuberances extending into the water column may keep cores oil free. Underwater photography may be important in this case.
- All cores need to be stored in insulated waterproof storage containers maintained at or near the lowest in-situ temperature. Remember that cores leak brine and will flush out even at very cold temperatures. Ideally, put cores into clean Teflon bags, then into a core tube, and finally into a cooler for transportation
- Make sure you maintain the orientation of the ice core during processing, storing, and labeling
- The same core can be used to take samples for biological and chemical analyses, only when there is sufficient material for both types of analyses, and when cores do not break during extraction

Note: The bottom of the ice can be very loose and will break up and drain while pulling the core up. This causes a significant loss of biological materials and oil. If this is the case, collect a surface water sample after removing the core to test for the presence of oil.

- On a single core per site, measure the temperature of the ice core immediately after coring:
  - Use the small drill to make a hole half way through the core. Insert the temperature sensor into the hole and take a reading. Once inserted, plugging the hole with the ice shavings adds an insulating layer aiding in temperature stability
  - Repeat this process over the entire ice thickness with a vertical resolution of 10 cm. If time allows, take greater resolution in the bottom 20 cm, at 1, 5, 10 15 and 20 cm from the ice-air interface to gain a better understanding of brine exchange with the underlying water. This may be important to understanding the degree of exchange with potential accommodated oil in the water.

In addition, ice core temperature allows for a greater understanding about brine volume fraction, ice porosity, etc.
  - Measuring the ice core temperature will allow you to make temperature adjustments during ice core storage and transportation to make sure that temperatures are kept as close as practical as the temperature of the ice. Remember that the colder the better to limit brine mobilization (non-biological sections)
- If possible, measure salinity from thawed ice samples using a handheld salinometer. Direct measurement of the brine presents specific challenges and might not give a representative salinity reading of the core sampled.
- After completing the temperature profile, store core samples as intact as practical. If space is limited or if indicated in the work plan, cut the ice core with an acetone-cleaned saw or knife into 2-10 cm long sections (depending on the total length), and place them in Teflon bags inside labeled core-storage boxes. These core segments can be used for biological or chemical analyses, noting that this was the core used for temperature profiles.
- In cores collected specifically for biological analyses:
  - Do not take a second temperature profile
  - Intact cores can be placed in Teflon bags inside labeled core-storage boxes, and stored for further processing in the laboratory (preferred)
  - Alternatively, cut cores into 2-10 cm sections (less ideal), place them in Teflon bags inside labeled core-storage boxes
  - If storage space is of concern, melt individual core section in 0.2 µm filtered seawater at 4°C and at the appropriate in-situ salinity, and concentrate fauna into 20 micron pore filter, and preserve with buffer formaldehyde (least ideal). As a rule of thumb, 1 cm of bottom ice (9-10 cm diameter corer) corresponds to 100 ml. Collect as many melted cores as practically feasible
- To sample ice plankton:
  - Collect a 7.5-10.5 cm diameter core and place it intact in a Teflon bag, inside of a labeled core-storage box, and stored for further processing in the laboratory. Depending on the length of the ice core, it may be necessary to segment the ice core with an acetone-cleaned saw or knife by cutting 2-10 cm long sections (or a longer section if specified in the work plan). Place each segment in individual Teflon bags inside labeled core-storage boxes until processed in the laboratory. Sectioning in the field reduces the spread of potential oil contamination as oil would likely leak from the brine channels oiling the entire length of the ice core
  - If sample storage space is a concern, melt the ice in surface water filtered through 0.2 µm polycarbonate membranes, recording the volume of water used (least ideal). If oil sheens or slicks are present, use reference surface water at the appropriate salinity and temperature (to avoid cell stress)
  - Melted ice samples for biological analysis should be preserved as follows:
    - For phytoplankton analysis with Lugol's solution followed by an acidified formalin solution (see Plankton guidelines)
    - For zooplankton analysis in 5% formaldehyde (preferred) or in 95% ethanol
    - Cap sample jars and place them a cooler, avoiding direct exposure to light
- Analyses performed in samples collected for biological metrics include:
  - Sea-ice flora:
    - Chlorophyll *a* profiles (via fluorescence) for biomass assessment and identification of species distribution within sea ice thickness (essential)
    - Sea-ice flora sample can be condensed by settling or by filtration throughout 10 micron pore filters and used for identification (species level) under light and electronic microscopy (suggested). Preserve samples by adding 38% formaldehyde at 1-4% final concentration (10% of total melted volume) and store in a glass or plastic vial until sorting
  - Sea-ice fauna:

    - Identification of species distribution within sea ice thickness, biomass, and abundance with focus on specific groups (i.e., nematode, a main component of cryopelagic community) (essential). If possible DO THIS LIVE as a method of pre-sorting as several taxa (e.g., the marine flat worms Platyhelminthes and Acoela) do not preserve well. Information can be gathered before preservation and more specific (species level) can be determined through observation or dissection
    - Reverse-flow filtration can be used to concentrate the invertebrate community throughout 20 micron pore filters to 5-10 ml volume, followed by preservation in 4 % formalin. These samples can be used for fauna enumeration using Bogorov's device and light microscopy (suggested). This type of analysis should be performed in a processing laboratory

Note: Most of the analyses for ice-flora can be performed in the field, but if time is of concern, samples can be processed in a laboratory as soon as practical

- To collect cores for chemical analysis:
  - Collect a 7.5-10.5 cm diameter core and place them intact in Teflon bags inside labeled core-storage boxes, and stored for further processing in the laboratory (preferred)
  - Alternatively, cut the core into approximately 3 cm sections (or other if specified in the work plan) (less ideal) and place cores in Teflon bags inside labeled core-storage boxes until processed in the laboratory. Care should be taken to keep track of the depth of each stored ice core section
  - Place sectioned ice cores in clean glass jar (amber preferred), cover the bottle with aluminum foil and leave at ambient temperature (only if ambient temperature is <5°C) until melted (least ideal). DO NOT expose the melted ice to ambient air. The jar must remain closed until processed in the laboratory
  - Make sure enough ice core sections are collected to meet volume requirements for chemical analyses
- For each ice core, record:
  - Date, time, and weather conditions (e.g., wind direction and speed)
  - Physical setting (shoreline orientation, exposure, etc.)
  - Ice core total depth, ice ore section depth range
  - Description of oiling conditions, using standard shoreline assessment terminology
  - Characteristics of the area surrounding the coring site: texture, color, biota, vegetation, debris, odor, presence of oil under the ice, etc.
  - Indicate if samples were melted with ambient temperature or filtered water
- If water samples are collected, wait ~10 min after coring to allow any disturbed under ice oil to dissipate in the area before taking samples.
- Discrete samples from a single sample point may be collected to represent a specific condition, such as a tarball for fingerprinting and source identification (see Stranded Oil guideline).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each ice core sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid, and ice core boxes. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)

  - Sample matrix (ice)
  - Sample #, date/time
  - Sampling method (drill). Note if sample is for QA/QC (field blank, trip blank, rinsate blank)
  - Ice oiling conditions (using standard shoreline assessment terminology), tidal elevation, weather conditions (e.g., wind direction and speed), ice characteristics, vertical changes in ice characteristics, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution on ice/under ice is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Keep all cores in coolers maintained at a temperature at or near the lowest in-situ temperature. Make sure storage procedures are sufficient to maintain the integrity of the cores. Refrigeration temperature shall be recorded upon sample storage, and monitored and recorded periodically to ensure proper refrigeration. Samples are to be excised from the core only at the processing laboratory.
- Immediately following collection, place all ice-melted samples collected for biological analyses in a cooler and keep at 6°C, and all ice-melted samples collected for chemical analyses in a cooler and keep at 4°C. DO NOT FREEZE. Store all samples in the dark. Refrigeration temperature shall be recorded upon sample storage and monitored and recorded periodically to ensure proper refrigeration.
- Do not use freshwater when sampling and preserving ice plankton samples.
- In below-freezing temperatures, collapsible jugs of warm water can be used in the cooler between ice-melted samples to prevent them from freezing.
- Water samples collected for chemical analysis can be held at 4°C in the dark for up to 7 days (includes recommended holding time in the field and receiving laboratory) without loss of sample integrity.
- THC and PAH: can add 1 mL of 6 N HCl/liter of sample within 2 hours of sampling to inhibit microbiological activity. Not required by EPA.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping. The receiving laboratory may provide packaging materials and shipping containers.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- Ship highly oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross contamination.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume and Requirements

Analytical Method	Sample Volume	Minimum Detection Levels [a]	Recommended Holding Time [b]	Minimum No. of Samples per Location [d]
BTEX (SIM)	1 liter [c]	0.1-1 µg/L	7 days	3 ice core replicates of at least 10 cm in length, or segmented ice cores per location; will depend on the total core depth
Total Hydrocarbons (THC) by GC/FID		15 µg/L		
PAH (including alkylated PAHs) by GC/MS-SIM		0.001-0.01 µg/L		
Chemical biomarkers (fingerprinting)		0.001-0.01 µg/L		

[a] µg/L= ppb; [b] Store at 4°C in the dark; [c] Several analyses can be made from a single sample; [d] 1 L of melted ice per sample type, 3 replicates per location– for biological analyses (see Plankton guidelines)

Analytical Methods

- Refer to those under Water and Plankton guidelines, if applicable

Key References

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Appendix A
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Ice Sample Collection Form

Sample Collection Form - ICE										
Lead Sampler's Name/Phone								Sampler Team Code		
Lead Sampler's Affiliation								Resource Group		
NRDA Contact/Phone								Resource Group Leader		
Incident Name								Habitat (e.g., ice)		
General Location Description								Sample date (mm/dd/yyyy)		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling Method**	**Sample Position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*Ice (I)*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Method of sampling (i.e., core)*	*Core depth*	*Core size and units*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD - YYY.YYYYYY*	*Description of sample, equipment used, photo numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										

Samples Relinquished by:				Received by:			
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball, Water, Snow, Ice, Sheen	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H) (SN)ow (I)ce (SH)een	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Complete sample IDs are comprised of the following information: Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Guidelines for Collecting Ephemeral Data in the Arctic: VEGETATED HABITATS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting data and samples from wet vegetated habitats (marsh-salt and brackish, inundated lowland tundra) during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Marshes in the Arctic are located in poorly drained soils and are periodically or continuously inundated by seawater. Freshwater marshes are not included as part of this guideline. Unlike marshes, inundated lowland tundra, composed of shrubs, grasses, mosses and lichens, is generally located in dry land, but it can be seasonally inundated or covered by wind-driven surge. Other vegetated habitats (e.g., eelgrass and kelp-boulder fields) are covered in separate guideline documents.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in vegetated habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate

Describe habitat

- Estimate the areal extent and degree of oiling on vegetation and on/in the substrate
- Document the presence/absence of biota and species composition
- Characterize the composition and health of plant communities and species

Study exposure

- Support exposure and transport modeling
- Document exposure to oil of biota in wet vegetated habitats

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical and biological characterizations

Collaboration

- Support other ongoing ephemeral sampling efforts (see Intertidal Sediment guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).

- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as a brackish marsh or inundated lowland tundra
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Modify this guideline as needed for marsh or inundated tundra habitats depending on the characteristics of the sampling area.
- Use a computer or conceptual model of the extent of vegetated habitat contamination or an appropriate power analysis to estimate the number of locations and number of sites per location needed to respond to the sampling objectives.
- If no other sampling design is indicated in the study plan, sampling can be performed using a stratified random sampling approach, which divides the sampling location into non-overlapping zones (strata) from which random samples are collected. This type of sampling improves the representative quality of samples by reducing sampling error.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) should be used to develop a sampling strategy and estimate distances, number of sampling transects, transect spacing, vegetated habitat width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with vegetation sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.

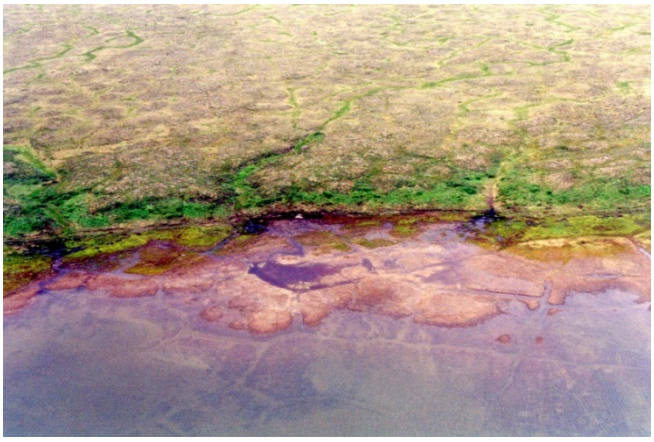
- Do as much material preparation prior to field deployment, including: labeling jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- Sampling of vegetation should be carried out in spring, summer or fall. Vegetation identification, characterization and sampling will not be possible in the winter as aboveground vegetation dies back and snow cover will make sampling difficult.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts on vegetated habitats.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended.
- Specific guidelines may be developed to aid in the sampling of specific endpoints or resources of interest within each vegetated habitat.
- The challenges of collecting samples in remote areas, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Areas for vegetated habitats include brackish and salt marshes and inundated lowland tundra.
- Sampling lowland tundra (see photograph) is particularly important if the oil spill coincides with periods of high wind-driven surge.
- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from oiled vegetated areas. Collecting pre-oiled vegetated habitat samples from vegetated



areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled "control" areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- Vegetation samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency is a function of oil persistence, biological community composition, habitat importance, and resource availability and should be defined in the study design.
- Stratify sampling locations by degree of oiling (Heavy, Moderate, Light), vegetation community type, and physical setting (exposure to wave energy, substrate type, proximity to freshwater sources, etc.).
- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting vegetation samples is at least two samples per location of relatively uniform oiling exposure. If relevant data are available, a power analysis or other modeling approaches should be used to determine the number of samples needed before going into the field.
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration
- Vegetation samples can be collected in conjunction with intertidal and stranded oil sampling.
- Close collaboration and coordination with other ongoing ephemeral data sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.
- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Instrument to measure physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, photosynthetically active radiation (PAR), and irradiance
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for sediment sampling under extreme cold conditions
- Zip lock bags – for non-chemical analysis of vegetation specimens
- 8 oz certified organic-clean jars with Teflon-lined lids and labels – for oiled vegetation samples
- Surveying supplies – 1.5 m stakes (two per transect), hammer or mallet to drive stakes in, profile rods (two, 1.5 m long, 2 cm intervals), 30 m fiberglass tape measure marked in cm, quadrats (0.25 m^2 and 1 m^2), hand level, shovel, and/or coring device, graduated probe to measure depth to permafrost
- FluorPen FP 100 handheld fluorometer with GPS module or equivalent with extra batteries – for fluorescence measurements (optional)
- Laminated sheets with species-specific fluorometer settings – for calibrating the handheld fluorometer (optional)
- Minolta SPAD-502+ handheld chlorophyll meter or equivalent with extra batteries – for Chlorophyll content measurements (optional)
- Pencils, waterproof pens, waterproof labels, markers, waterproof calculator
- Field Sample Forms (template in Appendix A)

- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- GPS, digital camera (with spare batteries), and photo scales
- Shoreline terminology code list and shoreline assessment survey guidelines (see Stranded Oil guidelines)
- Field sample form (plant identification field guides/charts, percentage estimation charts, vegetated habitat oiling datasheets), field notebook (waterproof paper), shoreline oil terminology code list, other guidelines as needed (Subtidal and Intertidal Sediment guidelines, Shoreline Assessment Survey guidelines)
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Sample labels
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment are not available):

- Sufficient quantities of pre-cleaned or disposable single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools (e.g., pre-cleaned stainless steel spoons).
- The only equipment to be used between sites is a shovel, which should be cleaned with soap and clean water. Repeated digging in clean sediments can be a last resort for cleaning the shovel if soap or

clean water are not available. Alternatively, use a clean dry towel or other dry material to clean the shovel before its next use. Additional cleaning may be required when working at oiled sites (see below).

- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples collected for chemical analysis:
  - Wash sampling equipment with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination of the site from oil on boots and shovels. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.
- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts, and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.

Study Design Implementation

- If recent SCAT data are available, it may not be necessary to conduct and overflight or ground survey. Otherwise, if practical, conduct an overflight of the affected area to locate and describe vegetated habitats, and generally characterize the degree of oiling on the shoreline. Note that weather, particularly fog, may dictate survey times that may not coincide with tides. Tides may not be a consideration along high Arctic shorelines where tidal range is small and water level is primarily wind driven.
- If an aerial survey is not feasible, survey from the ground. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs can be geo-referenced later. Observations should include:
  - Presence of vegetation, extent, and physical setting (wave exposure, ice scour, water circulation, proximity to freshwater sources, etc.)
  - Approximate width and degree of oiling of the shoreline adjacent to vegetated habitats (if applicable)
  - Locations of access points, major landmarks, and potential ground-truth and reference stations
  - Locations and approximate lengths of oiled vegetation segments
  - Obvious vegetation zonation
- Select locations for ground surveys based on degree of oiling, vegetation types, and similar physical setting as determined from overflight observations or SCAT data.
- Schedule ground surveys so that the vegetated habitat is not covered by water.
- At each selected location, conduct a preliminary ground survey to determine the potential magnitude of vegetation injury. Make an estimate of the acres of vegetation with different degrees of oiling and disturbances from response actions, if any.
- Based on the study design and/or sampling strategy outlined before going into the field, at each location establish a transect perpendicular to the shoreline from the upper edge of the vegetated

habitat to at least its seaward/aquatic extent and preferably to the water line. The transect placement should be representative of the habitat type and oiling conditions of the location.

- When establishing the transect:
  - Record the transect location using a GPS and accurately plot the transect location on a map or aerial photograph
  - If possible, permanently mark the transect location using "front" and "back" flexible markers driven into the ground that line up with the transect. Consider placement carefully to minimize loss due to vandalism, erosion, ice scouring during winter, etc. ***Note:*** Make the stakes short enough so that they are not a hazard to snowmobile traffic during winter
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one flexible marker is lost; note whether the angle reading is magnetic or corrected to true north
  - Take photographs of the transect at the beginning, middle, and end, including upslope, downslope, and alongshore images
- Measure the topographic profile along the transect. Make the transect intervals short enough to represent the topography of the site and changes in the plant community and oiling degree, but no longer than 3 m apart so that topography is accurately recorded. Profiles are useful due to the strong influence topography has on vegetation and oil distribution.
- For each interval along the transect, record:
  - Distance and elevation change
  - Oiling type description, thickness, and % cover on the substrate
  - Vertical oiling (total height of visual oil on vegetation; cm), oil thickness, and % oil cover on the vegetation
  - Depth of oil penetration into the sediments
  - Sediment type and grain size
  - Plant species present in the interval (be sure to delineate boundaries between plant communities)
  - Plant health (death or discoloration)
  - Type and extent of vegetation or substrate disturbance such as trampling, cutting, erosion, etc.
  - Presence, condition, and/or altered behavior of visible biota such as snails, worms, etc.
  - Presence of snow or ice
  - Depth to permafrost (if applicable)
  - At representative sites along the transect measure recommended physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, PAR, and irradiance. Follow instrument manual descriptions
- Collect samples from each site (described below) and record the distance along the transect and GPS coordinates of each sampling site.
- Take pictures of the transect before and after sampling and pictures of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Depending on the characteristics of eelgrass beds, cameras with underwater capabilities may be required (see below). Make sure each photograph or series can be later associated with the corresponding sampling station (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The

numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).

- At each selected sampling site, conduct a preliminary visual survey of the shoreline and draw a field sketch showing:
  - Shoreline orientation, type, grain size, etc.
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Location and general extent of the vegetated habitat
- Prioritize sample collection from each site based on logistics, sample storage, etc. as follows (in order of importance):
  - Estimation of percent vegetation cover and vegetation oiling
  - Description of vegetation community composition
  - Collection of infauna, invertebrates, and tissue samples
  - Collection of sediment, source oil, or other types of non-specific vegetation habitat samples
- If time allows, conduct detailed sampling to characterize impacts on vegetation using quadrats as follows:
  - Divide the width of the oiled vegetation along the transect in half, to an "outer" and "inner" band. Within each band, randomly select two locations to place a 1 m^2 quadrat. One approach to random location selection is as follows:
    - Place two tape measures at a 90° angle: 1) along the transect and 2) within each band
    - Use a random numbers table to randomly select two numbers
    - Place the center of the quadrat at the intersection of the two numbers on the tape measures
  - Photograph the quadrat from as vertical position as practical; include a photo scale and quadrat label in the photograph
  - Visually estimate live and dead vegetation coverage within 1 m^2 quadrat, including percent live vegetative cover by species, percent dead vegetative cover (not differentiated by species)
  - Visually estimate vegetation oiling impact index by oiling impact category:
    - No impact - vegetation appears to look natural, no evidence of oiling
    - Trace impact - vegetation shows minor chlorosis (yellowing of leaves), no evidence of oiling
    - Light impact - vegetation shows considerable chlorosis (<50% chlorosis), oil present on plant stems but variable on ground
    - Medium impact - vegetation shows chlorosis on >50% of leaves and stems, oil present on plant stems and ground
    - Heavy impact - vegetation dead, oil present on plant stems and ground
  - Measure apparent oiling height (in cm) on the tallest stem within the quadrat
  - Measure depth to permafrost just outside the quadrat
  - Measure water salinity and temperature if vegetation is inundated
  - If required by the study design, harvest all live stems within a 0.25 m^2 subsample for laboratory measurement of stem height, density and aboveground biomass by species. Consider that Arctic vegetation grows and recovers very slowly from disturbances and should not be harvested unless necessary. Place harvested material into high strength plastic bags, appropriately labeled and tied shut. Keep cold (~4°C) until processed
  - Sample three leaves from the tops of three plants of the same species for laboratory chlorophyll content measurements. Store these samples as described earlier. If available, hand-held chlorophyll meters can be used to determine chlorophyll content in the field
  - If time and resources allow, and if the proper training and knowledge are available, the following metrics could be collected:

    - Fluorescence measurement of damage to the photosynthetic apparatus of a plant leaf section with a FluorPen FP 100 handheld fluorometer with GPS module. Follow guideline provided in the instrument manual. Measure fluorescence on at least 3 unoiled leaves (1 to 3 leaves from the top) from at least 3 plants. Use laminated sheets to adjust the instrument's species-specific settings for actinic light (ultraviolet sunlight), if available, and measuring light levels
    - Chlorophyll content measurement (an indicator of plant growth status) using a Minolta SPAD-502+ handheld chlorophyll meter or equivalent on at least 3 leaves (1 to 3 leaf from the top) from at least 3 plants. Follow guideline provided in the instrument manual. If time or other factors are a concern, collect leaves for laboratory measurements, and follow the storage procedures described earlier
- Photo document each location (transect, transect intervals, quadrats, sample, etc.) (see Field Photography guideline). All photographs or videotape should be described in the photo log, located on a simple sketch map of the site, and noted on the data sheet. Make sure each photograph or series can be later associated with the location and specific sites (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline). Photographs or videotapes should include:
  - The general location and setting, showing permanent stakes
  - Examples of plant zonation and condition along the transect
  - Locations of quadrats and samples
  - Representative examples of the extent and degree of oiling
  - The extent and degree of trampling, burning, or other disturbance in the area
- Collect oiled vegetation for fingerprinting the source or quantifying the oil loading. Place oiled samples in a pre-cleaned (labeled) glass jar and place on ice in a cooler while in the field. Transfer to a freezer at the end of each day. Vegetation samples for confirming plant species identifications should be collected from unoiled areas, if possible, and can be placed in labeled plastic bags.
- To take sediment samples and samples of representative invertebrate species for tissue analysis, see Sediment Intertidal and Shellfish Tissue guidelines.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each vegetation sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (vegetation, sediment, tissue)
  - Sample #, date/time
  - Note if sample is for QA/QC (rinsate blank)
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.

- Documenting oil distribution in vegetated habitats is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments.
- Make a quick sketch in a field logbook or sketch form showing the sampling locations in enough detail that the location could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Immediately place all vegetation samples in a cooler and keep at approximately 4°C. Use frozen gel packs to maintain the temperature if ambient temperatures are above freezing. In below freezing temperatures, collapsible water jugs filled with warm water can be used to maintain the temperature if heated storage space is not available. A programmable temperature logger or thermometer should be placed in each cooler to maintain a record of storage temperatures.
- Protect samples from direct sun exposure (e.g., UV radiation).
- Tape lids on sample bottles so that they do not accidentally come off.
- If possible, store samples from unoiled locations in one set of coolers, with oiled samples in a separate set of coolers.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Freeze samples for chemical analysis as soon as practical or by the end of each day
- Vegetation samples for chlorophyll content analysis, species identification, plant characterization or other purposes should be maintained at 4°C. DO NOT FREEZE.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Analytical Methods

- Refer to those under Stranded Oil guidelines

Key References

Dalby, D.H. 1987. Salt marshes, Chapter 3: in J.M. Baker and W.J. Wolff (eds.), Biological Surveys of Estuaries and Coasts, Estuarine and Brackish Water Sciences Association Handbook, University of Cambridge Press, Cambridge, UK. pp. 38-80.

Hester, M. and J. Willis. 2010. Techniques and Analyses Suggested for Employment in Assessing Oil Impacts to Coastal Salt Marshes in the Gulf of Mexico. Shoreline Technical Group. NRDA, Deepwater Horizon Oil Spill. 22 pp.

NOAA Damage Assessment Center. 1997. Field forms and codes. Appendix 6: in Natural Resource Damage Assessment Emergency Guidance Manual, Version 3.1. NOAA Damage Assessment Center, Silver Spring, MD.

Appendix A
Supporting Documentation- Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:
- Vegetation Field Data Sheet (2 parts): General information, Transect data

Vegetation Field Data Sheet - General information (one per Location)

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder/ Affiliation: _____

3 Other Team Members / Affiliation: _____

4 **Site Descriptors**

Site Name/ID: _____

Latitude: _____ Longitude: _____

Habitat setting (check one): Supratidal _____ Intertidal _____ _____

Vegetated habitat Size: Width (m) _____ Length (m) _____

Overall vegetated habitat condition: _____

Vegetation percent coverage (%): _____

5 **Physical/Chemical Parameters**

Salinity (ppt): _____ Air temperature (C): _____

Weather/Cloud cover: _____

PAR (uEm^{-2}s^{-1}): _____ Oiling Condition (check 1): _____ None

_____ Sheen

_____ Light

_____ Moderate

_____ Heavy

6 **Sketch**

Drawing Legend	
~~	Water line
▬▬▬	Vegetation
----------	Transects

Additional information:

7 **Photos:**

Number	Description	Number	Description

Notes

Vegetation Field Data Sheet - Transect data **Page 1 of 3**

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other Team Members / Affiliation: _____

4 Transect ID: Transect Length (m):

	Waypoint #:	Latitude (DD)	Longitude (DD)	Transect Angle
Front Stake				
Back Stake				

Photos:	Number	Description	Number	Description

5 **Sketch**

Drawing Legend	
FS	Front Stake
BS	Back Stake
~~	Water line
▬▬▬	Vegetation bed
----------	Transects
☐	Quadrat
V	Plant Sample
S	Sediment Sample
W	Water Sample
I	Invert. Sample

6 **Transect Characterization**

Distance (m)	Change in elevation (+ or - cm)	Vegetation cover (%) by species if possible	Plant health (e.g., chlorosis, necrosis)	Oil Descriptors			Sediment grain size/ Depth of permafrost	Substrate disturbance (describe)
				Vegetation (Height, thickness, % Cover)	Substrate (Type, Thickness, % Cover)	Depth of Penetration (cm)		

Notes:

7 Quadrat Characterization

Quadrat (Inner/ Outer)	Distance from transect (m)	Oiling Impact Index[1]	Vegetation Cover (%)		Species Measured	Chlorophyll	Fluorescence	Sample taken (yes/no)
			Live	Dead				

[1] **Vegetated Habitat Oiling Impact Index:**
1 - None - vegetation appears to look natural, no evidence of oiling
2 - Trace - vegetation shows minor chlorosis, no evidence of oiling
3 - Light -oil present on veg. but variable on sediment, chlorosis on <50% of leaves
4 - Medium - oil present on vegetation and sediment, chlorosis on >50% of leaves
5 - Heavy - vegetation dead, oil present on plant stems and sediment

8 Point Sample Collection (use appropriate field data sheets for point samples)

Vegetation samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Vegetation samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for grain size analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Water samples for chemical analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Invertebrate samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Tissue samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Other samples

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Guidelines for Collecting Ephemeral Data in the Arctic: EELGRASS HABITATS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting of samples from eelgrass habitats for chemical and biological analyses during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Eelgrass in the Arctic occurs along the Bering Sea coast from the Alaska Peninsula and eastern Aleutian Islands to the Bering Strait, and in the southern Chukchi Sea on the Seward Peninsula to Kotzebue Sound. Kotzebue Sound, inside Cape Espenberg is the northern limit of eelgrass in Alaska. Eelgrass beds are mostly, if not exclusively, found in lagoons or bays. Eelgrass occupies the lower intertidal along the Alaska Peninsula, and they are exclusively subtidal north of the Yukon-Kuskokwim Delta. Information about the occurrence of eelgrass can be found in ESI maps and ShoreZone data.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in eelgrass habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate
- Characterize other sources of oil or hydrocarbons in the environment

Describe habitat

- Estimate the areal extent and degree of oiling in eelgrass habitats
- Document the presence/absence and species composition of the vegetation, and estimate semi-quantitatively the abundance or density of the eelgrass-associated invertebrate community

Study exposure

- Measure oil related compounds in biological tissues
- Support exposure and transport modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of chemical and biological characterizations

Collaboration

- Support other ongoing efforts (see Subtidal Sediment guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).

- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.
- Safety is of primary importance when sampling in the Arctic. If snorkeling or use of Self Contained Underwater Breathing Apparatus (SCUBA) is consider for subtidal eelgrass sampling, ensure that personnel with underwater sampling experience (for snorkeling) or a certified dive team leader (for SCUBA) are part of the sampling team (See Appendix A for general guideline on Snorkeling/SCUBA safety).

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of eelgrass habitat impacts or an appropriate power analysis to determine the number of sampling locations needed to respond to the sampling objectives.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- Shoreline visualization tools (e.g., ESI maps, satellite images, ShoreZone) and traditional ecological knowledge should be used to determine where eelgrass beds are present, and to develop a sampling strategy and estimate the number of sampling transects, transect length and spacing, intertidal zone width, etc. before going into the field.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with eelgrass sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels), etc.

- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts on eelgrass habitats.
- When sampling in remote areas with limited shipping capabilities, plan sampling trips lasting no longer than three consecutive days, then bring all samples to the main facility for shipping. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.
- Samples from eelgrass habitats can be collected from boats, by wading in shallow water, or by SCUBA or snorkeling in subtidal areas (e.g., north of the Yukon-Kuskokwim Delta). Safety training would be required when sampling via SCUBA.
- Snorkeling/SCUBA activities require extensive coordination with other ongoing spill operations. Snorkeling/SCUBA activities are not to interfere with response operations, and are to be avoided in areas with heavy on-water/boating traffic.

Area selection

- Eelgrass is a sensitive and biologically productive habitat, so sampling the discreet areas where it occurs should be a very high priority for ephemeral data collection.
- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other sources to determine what areas have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- Collect background samples from areas representative of pre-oiling conditions, as well as areas not yet oiled but in the potential path of the oil. These data will provide the best evidence of changes due to exposure to the spilled material. These samples should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Samples from eelgrass habitats should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter.
- The number of locations and number of transects per location should be defined in the study design. At a minimum, survey 3-5 control/least oiled transects and 3-5 oiled transects by eelgrass zone

(upper, middle, lower eelgrass bed edge). If oiling is relatively uniform across the eelgrass bed, survey at least 3 transects, or enough to characterize the eelgrass bed. If relevant data are available, a power analysis or other modeling approaches should be used to determine the number of transects and sites per transect needed before going into the field. If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one reference/pre-oiled location and two heavily oiled locations.

- Sample along exposure gradients, starting in the cleanest zone and at regular intervals proportional to the exposure area.

Collaboration

- Samples from eelgrass habitats can be collected in conjunction with sediment and water sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the study plan, desired sampling capacity, and logistics. See Alternative equipment/methods guideline for options in preferred equipment is not available.

- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs – for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Instrument to measure physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, turbidity (Secchi disk), photosynthetically active radiation (PAR), and irradiance
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for water sampling under extreme cold conditions
- Zip lock bags – for non-chemical analysis of vegetation specimens
- 8 oz. organic-cleaned (solvent rinsed) glass jars – for sediment and oiled vegetation samples
- Transect supplies – flexible markers (two per transect), hammer or mallet to drive markers in, profile rods (two, 1.5 m long, 2 cm intervals), 30 m fiberglass tape measure marked in cm, quadrats (0.25 m^2, 0.1 m^2), hand level, shovel and/or coring device
- For invertebrate sampling: Airlift sampler with hoses, compressed gas tank and regulator; a 250 µm sieve – for airlift contents sorting; 10% formaldehyde in seawater – for invertebrate preservation; 70% ethanol – invertebrate preservation for the purpose of identification and counting; and a stain (such as Rose Bengal) if appropriate
- Waterproof ruler and tape measure
- Sorbent pads
- SCUBA and snorkeling gear, including dive flag and weight belt (for subtidal eelgrass sampling)
- SCUBA underwater writing slate (with pencil and pencil with safety leash) and underwater notebooks (for underwater record keeping)
- Plastic snowshoes (for shallow water sampling)
- Sediment corer (for invertebrate sampling)
- Field Sample Forms (template in Appendix B)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- Field notebook (waterproof paper)
- Calculator
- GPS, digital camera with underwater capabilities (with spare batteries), photo scales

- High-resolution underwater video camera with GPS capabilities, underwater light and towfish (when other types of surveys are unsafe or unpractical)
- Shoreline terminology code list and shoreline assessment survey guidelines (see Stranded Oil guidelines)
- Sample labels
- Suitable disposal bags for oiled PPE and disposable sampling materials

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.
- Non-disposable sampling devices (air lift sampler) MUST be decontaminated between samples:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- If possible, dedicate one set of sampling equipment per degree of oiling to minimize potential cross-contamination. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal equipment should be exchanged or cleaned between sites if it becomes contaminated.
- Take precautions to avoid cross-contamination of the site from oil on boots and equipment.

Study Design Implementation

- If recent SCAT data are available, it may not be necessary to conduct and overflight or ground survey. Otherwise, if practical, conduct an overflight of the affected area within two hours of low tide (before or after), to locate and describe eelgrass beds and generally characterize the degree of oil exposure on the adjacent shoreline or the water surface over the beds.
- If an overflight is not feasible, observations can be made from boats. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs can be geo-referenced later. Observations should include:
  - Eelgrass bed presence, extent, and physical setting (wave exposure, ice scour, water circulation, proximity to freshwater sources, etc.)
  - Approximate lengths and degree of oiling of the shoreline adjacent to eelgrass beds

– Locations of access points, major landmarks, and potential ground-truth and reference stations

Note: Overflights may not be adequate enough to locate and identify subtidal eelgrass beds. It is highly advisable to consult with local experts and to select sampling sites based on trajectory modeling or similar.

- Upon return, select sites for ground surveys based on degree of oiling (heavy, moderate, light, no oil; standard SCAT terminology), bed size, and similar physical setting.
- Based on the study design and/or sampling strategy outlined before going into the field, establish a minimum of three transects spaced at least 30 m apart (recommended) (Figure 1). When sampling intertidal eelgrass beds, transects should be perpendicular to the shoreline and encompass the entire intertidal zone. Run transects within two hours of low tide (before or after). Tides are only a consideration south of Bering Strait where the tidal range (other than storm tides) is about 1 m. In the Chukchi the range is <30 cm. Snorkeling or underwater observations may be required to identify the outer edge of the eelgrass bed. When sampling subtidal eelgrass beds, transect should be perpendicular to the shoreline and encompass the width of the eelgrass bed.

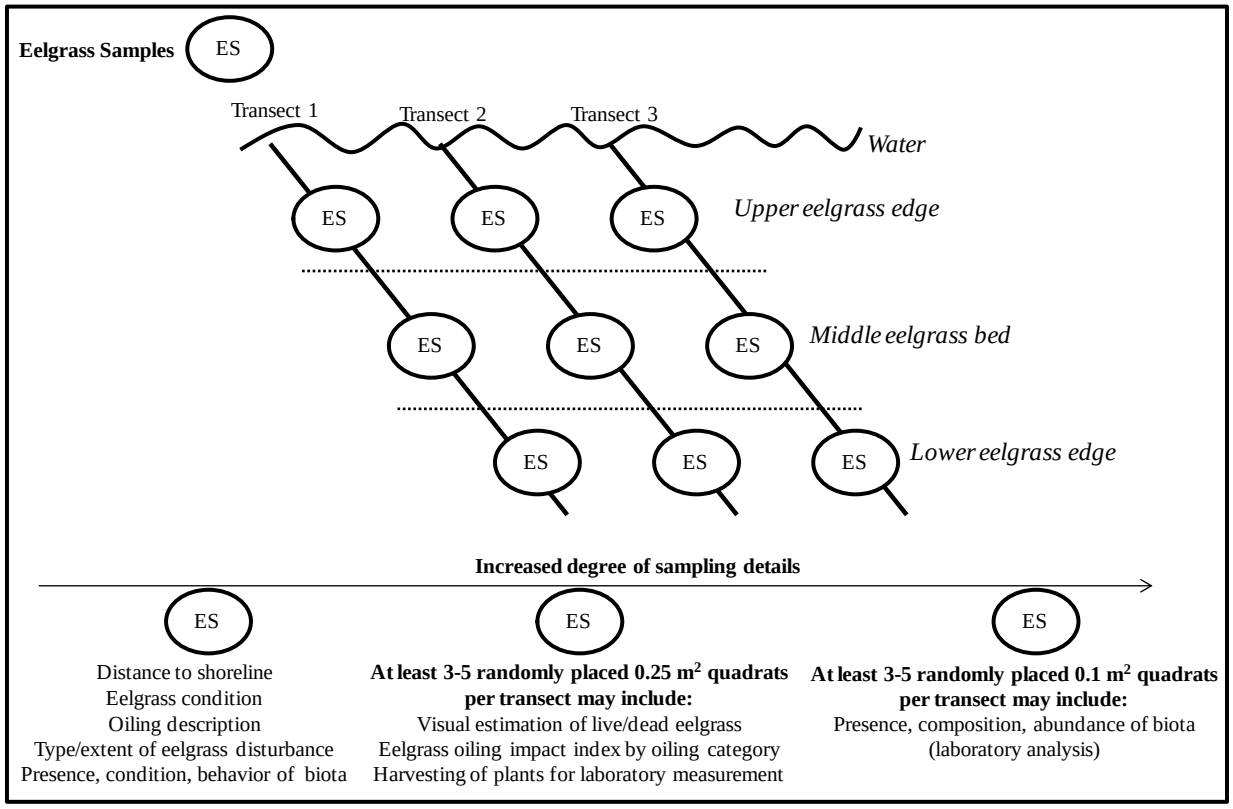


Figure 1. Schematic representation of the recommended intertidal eelgrass sampling strategy, including transects and sampling sites. Subtidal eelgrass sampling strategy not shown. Dashed lines represent approximate tidal zones. ES = eelgrass sample. Area-specific modifications may be needed.

- When establishing transects:
  - Record the transect location using a GPS and accurately plot the transect location on a map or aerial photograph
  - If possible, permanently mark the transect location using "front" and "back" flexible markers attached to the bottom/substrate that line up with the transect. Consider placement carefully to minimize loss due to vandalism, erosion, ice scouring during winter, etc.
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one flexible marker is lost; note whether the angle reading is magnetic or corrected to true north

  - Take photographs of the transect at the beginning, middle and end, including upslope, downslope, and longshore images. This takes little time and establishes a reference for future work

Note: Make the stakes short and visible so that that they are not a hazard to people traveling by snowmachine or all-terrain vehicle.

- For each transect, record:
  - Date, time, weather conditions (e.g., wind direction and speed), and tide level (if applicable)
  - Physical setting (shoreline orientation, exposure, etc.)
  - Length of the transect (in meters) and of the sampling zone
  - General characteristics of the eelgrass bed condition including death or discoloration
  - Extent and degree of visible shoreline oiling (use SCAT standard terminology for a more detailed assessment of shoreline oiling if needed)
  - Vertical oiling (total length of visual oil on vegetation; cm), oil thickness, and % oil cover on the vegetation
  - Oiling type description, thickness, and % cover on the substrate
  - If possible, depth of oil penetration into the sediments, if any (using a shovel or coring device; applicable to intertidal sampling only)
  - Sediment type (grain size)
  - Type and extent of eelgrass or substrate disturbance such as trampling, cutting, erosion, etc.
  - Presence, condition, and /or altered behavior of visible biota such as amphipods, gastropods, crabs, etc.
  - Extent and degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Additional parameters that should be recorded are described in the field data sheet
- When sampling intertidal eelgrass beds, divide each transects into sampling intervals based on the extent of the bed such as upper edge, middle bed, and lower edge. When sampling subtidal eelgrass beds, divide each transects into sampling intervals of equal width.
- Collect samples from each site (described below) and record the distance along the transect and GPS coordinates of each sampling site.
- Take pictures of the transect before and after sampling and pictures of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix B. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Depending on the characteristics of eelgrass beds, cameras with underwater capabilities may be required (see below). Make sure each photograph or series can be later associated with the corresponding sampling station (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- A calibration exercise is needed to ensure that all field teams consistently perform eelgrass sampling.
- The use of plastic snowshoes is recommended when collecting samples at low tide, so not disturb the bottom sediments.
- At each selected sampling site, conduct a preliminary visual survey of the shoreline and draw a field sketch showing:

  - Shoreline orientation, type, grain size, etc.
  - Extent and degree of shoreline oiling (use shoreline oil terminology codes and % cover charts)
  - Type or degree of shoreline cleanup, if already performed (particularly note sediment disturbance or removal, flushing of oil and/or sediments into down-slope areas, etc.)
  - Location and general extent of the eelgrass bed
- Prioritize sample collection from each site based on logistics, sample storage, etc. as follows (in order of importance):
  - Estimation of percent vegetation cover and eelgrass oiling
  - Collection of eelgrass specimens
  - Collection of infauna, fish and invertebrates, and tissue samples
  - Collection of sediment, source oil or other types of non-specific eelgrass habitat samples
- At each sampling site, measure recommended physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, turbidity, PAR and irradiance. Follow instrument manual descriptions. If time is of concern, or if the appropriate instruments are not available, only take salinity and temperature measurements.
- When establishing quadrats at a site along a transect:
  - Take photographs of each entire quadrat from an angle as vertical as practical
  - If possible, describe the sediment composition and sediment oiling, including depth of oil penetration
  - Prior to sampling, note and describe any associated biota, including their behavior
  - For detailed sampling, if required in the sampling plan, place randomly 3-5 0.25 m^2 quadrats per transect and estimate:
    - Visual estimation of live and dead percent vegetation cover and record the number of flowering shoots
    - Visual estimation of eelgrass oiling impact index by oiling impact category:
      - No oiling - eelgrass appears to look natural, no evidence of oiling
      - Trace oiling - eelgrass shows minor necrotic lesions, no evidence of oiling
      - Light oiling - eelgrass shows considerable necrotic lesions on <50% of leaves, oil present on eelgrass but variable on sediment
      - Medium oiling - eelgrass shows necrotic lesions on >50% of leaves, oil present on eelgrass and sediment
      - Heavy oiling - eelgrass dead, oil present on plant stems and sediment
    - Sample aboveground vegetation by clipping intact shoots in 0.10 m^2 quadrants. These samples can be used to quantify shoot density and percent flowering shoots in the sample. The state of the flower/seed development should also be noted (phenology). Leaf length and width should also be recorded. Measure the length of the longest leaf in a shoot and its width. ***Note***: Quadrat size can be adjusted (typical range 0.04 to 0.25 m^2) based on the height and density of the eelgrass bed (e.g., smaller frames for denser beds)
    - Harvest at least 10 randomly selected plants with intact aboveground structures for laboratory measurement of biomass. Place harvested material into high-strength plastic bags, appropriately labeled and tied shut. Keep cold (~4°C) until processed
- Collect vegetation samples for chemical analysis from the quadrats, as needed. Oiled samples can be used to fingerprint the oil or measure the amount of oil on eelgrass, which can be used in mass balance analyses. Place oiled samples in a pre-cleaned glass jar and place on ice in a cooler while in the field. Transfer to a freezer at the end of each day. Follow vegetation collection guidelines and use appropriate quality control measures.
- If faunal organisms are observed, or if key species are expected to occur (ecologically important prey species, etc.), sampling can be conducted to document the presence, composition, and general abundance of organisms. If highly quantitative density estimates are needed, or detailed comparisons of oiled vs. un-oiled sites are planned, an experienced eelgrass ecologist and statistician should be

consulted to plan more detailed studies. For semi-quantitative surveys of selected shoreline transects or habitats:

- For species presence/absence, composition, or rough density estimates use quadrats (0.1 m^2). Randomly place the quadrat 1 and 2 m to the left and right of each transect at locations selected by water depth of the bed (upper, middle, lower eelgrass edge), for a n=4 at each station along the transect
- Vacuum all material within each quadrat to a depth of 10 cm using an airlift sampler:
  - Attach hoses and airlift sampler to the compressed gas tank equipped with a regulator
  - With the regulator closed, open the main tank valve all the way and turn the valve back one full turn
  - Slowly open the regulator until the pressure in the range of 20-45 psi (140-310 kPa)
  - Place the inlet of the airlift sampler on the quadrat substrate
  - Deliver short pulses of compressed gas to the sampler to suction the contents of the quadrat and approximately the top 5 cm of substrate
  - Filter contents through a 250 μ m sieve and transferred into sampling jars
  - Preserve samples in 10% formaldehyde buffer or in 70% ethanol (in sea water) until processed in the laboratory and stain (such as Rose Bengal) if appropriate
- Place a waterproof label with the station location, sample number, and date on the sample container

- Infauna (polychaetes, etc.) can be collected with a sediment corer by pushing the corer into the sediment and transferring contents to a sampling jar. Record the approximate length of the core.
- If tissue analyses are planned for larger infauna, such as bivalves, follow the Shellfish Tissue guideline. Briefly:
  - Wrap each individual specimen in pre-cleaned aluminum foil and freeze as soon as practical
  - If time allows, take photo of sample
  - Take care to avoid cross contamination during sampling and handling
  - Clean sampling equipment, such as shovels, between collections
  - Ship biological and sediment samples on ice overnight (ideally) to the laboratory conducting the analyses. Include Chain of Custody forms
- Mobile organisms including fish and invertebrates can be sampled from eelgrass habitats with a seine or a push net (see Fish guidelines).
- Take sediment samples and samples of representative invertebrate species for tissue analysis particularly from locations where eelgrass impacts are visibly oiled. If oiled locations are sampled, fauna from reference locations should also be collected (see Shellfish Tissue and Subtidal Sediment guidelines).
- If possible, store samples from unoiled locations in one set of coolers, with oiled samples in a separate set of coolers.
- If direct observations (intertidal transects, or surveys via SCUBA/snorkel) are unpractical or unsafe, consider using high-definition underwater videography deployed from vessels to provide documentation of percent vegetation cover and eelgrass oiling. This type of sampling requires previous training. Briefly:
  - Mount the underwater camera in a ‘down-looking’ orientation on a towfish deployed directly off the stern of the vessel
  - Allow the camera to follow the eelgrass contour above the canopy
  - Maintain the field of view as constant as practical (1 m^2)
  - The vessel speed should be held as constant as practical (about 1 m/s) to facilitate estimation of distances
  - Conduct straight line underwater video transects (randomly selected) perpendicular to the shoreline and encompassing the width of the eelgrass bed

  - Carefully catalogue all underwater videos and ship to the appropriate laboratory for processing and interpretation. Post-processing of underwater videos can be used to estimate eelgrass vegetation coverage
- Photo-document each site. All photographs or videotape should be described in the photo log, located on a simple sketch map of the site and noted on the data sheet. Photographs or videotapes should include:
  - The general site location and setting, showing permanent flexible markers
  - Examples of eelgrass distribution and condition along the transect
  - Locations where samples were collected
  - Representative examples of the extent and degree of oiling
  - The extent and degree of trampling or other disturbance in the area
  - Examples of services provided by the eelgrass habitat (animal use, shoreline protection, etc.)

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Complete the Chain of Custody form, noting where each sample was collected, sampling device used, time/date of collection, size and container type, and sampler name (see Chain of Custody guidelines).
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the following on the field sample form for each sample:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample #, date/time
  - Describe the oiling conditions (using standard shoreline assessment terminology), tidal elevation (if applicable), weather conditions (e.g., wind direction and speed)
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Documenting oil distribution in intertidal areas is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods. These data may be collected by SCAT teams and available to support environmental assessments. Samples may be needed for fingerprinting or monitoring weathering, to correlate a degree of oiling term with oil loading, to confirm the presence of oil, or for bioassay purposes.
- Make a quick sketch in a field logbook or sketch form showing the sampling stations in enough detail that the station could be re-occupied by someone else.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Tape lids on sample bottles so that they do not accidentally come off.
- Ship known oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross-contamination.
- Ship any samples preserved in formalin or other chemicals as hazardous goods. Include Chain of Custody forms.
- Refrigeration temperature shall be recorded upon sample storage, and monitored and recorded periodically to ensure proper refrigeration.

- Use packing material, such as bubble wrap, around containers to prevent breakage during handling and shipping.
- Ship samples directly to the laboratory as soon as practical with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended storage temperatures.

Analytical Methods

- Refer to those under Shellfish Tissue and Subtidal Sediment guidelines.

Key References

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Appendix A
General Guideline for Snorkeling/SCUBA Sampling of Nearshore/Subtidal Eelgrass Beds

The guideline below provides a series of recommendations to ensure the safety of underwater sampling. These recommendations are not intended to be comprehensive. Consult additional sources of information on underwater safety. Remember that the success of underwater operations result from careful planning and observation of safety precautions.

- If underwater hazards are identified (e.g., strong currents, unsafe wave conditions), cancel or suspend any underwater sampling until the hazard is no longer an issue.
- Snorkeling/SCUBA diving are not recommended when oil slicks are present at the water surface; however, it may be acceptable if only light sheens are present.
- Snorkeling/SCUBA activities are NOT to interfere with response operations.
- Night snorkeling/SCUBA is not recommended.
- When planning underwater activities follow these recommendations:
  - Have all emergency information and phone numbers handy
  - Inspect all essential underwater equipment prior to initiation of underwater activities
  - Establish a communication system between underwater samplers, and between underwater samplers and personnel at the surface
  - Document all planned depths and sampling type (snorkeling; SCUBA)
  - Be aware of your surroundings and consider environmental conditions, currents, tides, visibility, air/water temperature, accessibility, and other natural and man-made hazards
  - Be aware of other activities in the area that may interfere with the dive or that pose a safety hazard (i.e., vessel traffic, noise, pollution, etc.)
  - Thermal protection should be ensure at all times by having wet or dry suits with hoods available. Dry suits are recommended for sampling in waters below 50°F, or when sheen is observed at the water surface.
  - Remember that because cold affects dexterity, underwater activities in cold waters must be shorter those in warmer waters
  - Sampling team members should be trained in cardiopulmonary resuscitation (CPR) and first aid, and should be able to recognize and treat any signs of hypothermia
- Specifically for SCUBA:
  - All divers must be properly trained on safe underwater procedures
  - Ensure that there are enough air tanks available for the duration of sampling activities
  - Maintain a Dive Log
  - SCUBA requires a 2-buddy system and should remain in visual sight of each other during ingress/egress and while underwater
  - A warning flag must be displayed at the dive location
  - Document and report any incidents that occurred during underwater sampling

Appendix B
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on waterproof paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:

- Eelgrass field Data Form – 2 parts: General information, Transect data

Eelgrass Field Data Form - General information (one per bed)

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder/ Affiliation: _____

3 Other team members / Affiliation: _____

4 Site Descriptors

Site Name/ID: _____

Latitude: _____ Longitude: _____

Habitat setting (check one): _____ Intertidal _____ Subtidal - Depth (m): _____

Bed Size: Width (m) _____ Length (m) _____

Overall bed condition: _____

Eelgrass percent oil coverage (%): _____

5 Physical/Chemical Parameters

Bottom salinity (ppt): _____ Air temperature (C): _____

Bottom temperature (C): _____ Bottom dissolved oxygen (mg/L): _____

Weather/Cloud cover: _____

Wave height (m): _____ Secchi depth (cm): _____

PAR ($uEm^{-2}s^{-1}$): _____ Oiled Condition (check 1): _____ None

_____ Sheen

_____ Light

_____ Moderate

_____ Heavy

	Irradiance	Depth (m)
Value 1		
Value 2		
Value 3		

6 Sketch

Drawing Legend	
~~	Water line
▬▬▬	Eelgrass bed
----------	Transects

Additional information:

7 Photos:

Number	Description	Number	Description

Notes

Eelgrass Field Data Form - Transect data (one per transect)

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliation: _____

4 Transect ID: Transect Length (m):

	Waypoint #:	Latitude (DD)	Longitude (DD)	Transect Angle
Front Flexible Marker				
Back Flexible Marker				

Photos:

Number	Description	Number	Description

5 Sketch

Drawing Legend	
FM	Front Flexible Marker
BM	Back Flexible Marker
~~	Water line
▬▬▬	Eelgrass bed
----------	Transects
☐	Quadrat
V	Plant Sample
S	Sediment Sample
W	Water Sample
I	Invertebrate Sample

6 Transect Characterization

Interval Length (m)	Eelgrass cover (%)	Eelgrass condition (e.g., chlorosis, necrosis)	Flowering shoots (yes/no)	Oil Cover (%)		Sediment type (grain size)	Oil penetration depth (cm)	Substrate disturbance (describe)
				Eelgrass	Substrate			

Notes:

7 Quadrat Characterization

Quadrat Location	Dist. from transect (m)	Cover (%)		Sample taken (yes/no)	Eelgrass and associated species	Count	Sample taken (yes/no)
		Live	Dead				

[1] **Eelgrass oiling impact scale:**

1 - None - eelgrass appears to look natural, no evidence of oiling

2 - Trace - eelgrass shows minor necrotic lesions, no evidence of oiling

3 - Light -oil present on eelgrass but variable on sediment, necrotic lesions on <50% of leaves

4 - Medium - oil present on eelgrass and sediment, necrotic lesions on >50% of leaves

5 - Heavy - eelgrass dead, oil present on plant stems and sediment

8 Point Sample Collection (use appropriate field data sheets for point samples)

Vegetation Samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Vegetation Samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for grain size analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Water samples for chemical analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Invertebrate samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Invertebrate samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Other samples

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Guidelines for Collecting Ephemeral Data in the Arctic: KELP-BOULDER FIELD HABITATS

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guidelines Objectives

The primary objective of this document is to provide guidelines on collecting of field data and samples from kelp-boulder field habitats for chemical and biological analyses during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. Dense kelp fields grow in a few areas in the Beaufort Sea, most notably the Boulder Patch (3-6 m deep) behind the barrier islands of Stefansson Sound, and sparse areas of kelp occur in Camden and Demarcation Bay. Boulder patches occur on the Bering Sea shelf northward to high Arctic and Beaufort Sea. There are few kelp fields in the Chukchi Sea, mostly located nearshore or in coastal lagoons. One notable field occurs in Peard Bay area by Barrow. These fields are local hotspots of abundant and diverse marine life where boulders provide rare hard substrate for macroalgae, epilithic invertebrates, and a variety of epibenthic macrofauna, which include motile invertebrates and fish. It should be noted that while kelp/boulder habitats are found at various locations across the Beaufort, they cannot be compared or used as replicates/controls for each other as it is known that the communities are very different. Alaska is home to three canopy kelp species: *Macrocystis pyrifera* (giant kelp), *Nereocystis luetkeana* (bull kelp), and *Eualaria fistulosa* (dragon kelp). There are many understory kelp species in Alaska, but species of note in the High Arctic are: *Alaria marginata* (winged kelp), *Laminaria solidungula* (Arctic suction-cup kelp), and *Saccharina latissima* (sugar kelp). This guideline may be applicable to sampling kelp habitats in the Aleutians and other areas outside the high Arctic; however, modifications would be needed. More appropriate guidelines for sub-Arctic and temperate areas may be available.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in kelp-boulder field habitats compared to background concentrations
- Determine the source of contamination via chemical fingerprinting analysis and characterize oil weathering and fate

Describe habitat

- Estimate the areal extent and degree of oiling on kelp-boulder fields
- Document the extent and duration of exposure to the spilled material and its bioavailability
- Document the presence/absence and species composition, and to estimate the abundance or density of the kelp-boulder field-associated invertebrate community

Study exposure

- Measure oil-related compounds in biological tissues
- Support exposure and transport modeling

Quality assurance/quality control

- Ensure the integrity of the sample(s) throughout sampling, transport, and storage

- Ensure the reliability of chemical and biological characterizations

Collaboration
- Support other ongoing efforts (see Subtidal Sediment guidelines)

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.
- Safety is of primary importance when sampling in the Arctic. Because Self Contained Underwater Breathing Apparatus (SCUBA) is the only way kelp-boulder fields can be sampled, ensure that personnel with underwater sampling experience or a certified dive team leader are part of the sampling team (See Appendix A for general guideline on SCUBA safety).
- Snorkeling/SCUBA activities require extensive coordination with other ongoing spill operations. Snorkeling/SCUBA activities are not to interfere with response operations, and are to be avoided in areas with heavy on-water/boating traffic.

Study design
- It is important to have a defined sampling strategy prior to conducting fieldwork.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an kelp field or boulder patch
  - Transect = a line through a site along which samples are collected or observations are made
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Use a computer or conceptual model of the extent of kelp-boulder field contamination or an appropriate power analysis to estimate the number of sampling locations and number of samples per site needed to respond to the sampling objectives.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with kelp-boulder field sampling. Tarballs, sheens, or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment
- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.

- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- Consider area-specific conditions for remote Arctic regions and make adjustments in methodology and equipment as necessary.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); solvent rinsing of jars and aluminum foil for sample storage, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations in the spread of oil over known kelp-boulder fields.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.
- Kelp-boulder field samples can only be collected by SCUBA. Safety training is required when sampling via SCUBA and special considerations will be required for sampling in contaminated water.

Area selection

- Sampling locations should include kelp-boulder fields have been or may be oiled.
- Use trajectory models, conceptual models, overflight information, SCAT data or other tools to determine what kelp-boulder fields have been exposed to oil and which ones are likely to be exposed.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It is a high priority to collect samples from oiled kelp-boulder field areas and pre-oiled kelp-boulder field areas that are likely to be oiled by the spill in the near future.
- Faunal samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency is a function of oil persistence, biological community composition, habitat importance, and resource availability and should be defined in the study design.

- The number of locations and number of sites per location should be defined in the study design. A minimum guideline for collecting kelp-boulder field samples is three per location of relatively uniform oiling exposure (see below).
- Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area.

Collaboration
- Samples from kelp-boulder fields can be collected in conjunction with intertidal and subtidal sediment and water sampling.
- Water and sediment samples should be collected to document oiling conditions in kelp-boulder fields habitat (see Water and Subtidal Sediment guidelines).
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the study plan, desired sampling capacity, and logistics. See Alternative equipment/methods guideline for options in preferred equipment is not available.
- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Instrument to measure physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, turbidity (Secchi disk), photosynthetically active radiation (PAR), and irradiance
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Zip lock bags– for vegetation specimens
- Transect supplies – 4 ft. stakes (two per transect), lead line or stainless wire (coated is preferable) use as permanent transect line that can be left in the field, hammer or mallet to drive stakes in, profile rods (two, 1.5 m long, 2 cm intervals), 30 m fiberglass tape measure marked in cm, quadrats (sizes ranging from 0.25 to 1 m^2), coring device, air-lift sampler
- For invertebrate sampling: Airlift sampler with hoses, compressed gas tank, and regulator; a 250 µm sieve − for airlift contents sorting; 10% buffered formalin (preferred), 95% ethanol (less ideal) − for invertebrate preservation; and a stain (such as Rose Bengal) if appropriate
- Waterproof ruler and tape measure
- Sorbent pads
- SCUBA gear, including dive flag and weight belt
- SCUBA underwater writing slate (with pencil and pencil safety leash) and underwater notebooks (for note taking)
- Sediment corer (for invertebrate sampling)
- Field Sample Forms (kelp-boulder oiling datasheets, percentage estimation charts– consider using underwater forms or waterproof paper; template in Appendix B)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guideline)
- Field notebook (waterproof paper), other guidelines as needed (Field Photography and Subtidal Sediment guidelines)
- Calculator
- GPS (waterproof), digital camera with underwater capabilities (with spare batteries), photo scales
- High-resolution underwater video camera with GPS capabilities, underwater light and Towfish (when other types of surveys are unsafe or unpractical)

- Shoreline terminology code list and shoreline assessment survey guidelines
- Sample labels
- Suitable disposal bags for oiled PPE and disposable sampling materials

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when processing samples for chemical analysis and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.
- Non-disposable sampling devices (air lift sampler) MUST be decontaminated between samples collected for chemical analysis:
  - Wash sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used
  - Rinse with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- If possible, dedicate one set of sampling equipment per degree of oiling to minimize potential cross-contamination.
- Take precautions to avoid cross-contamination of the site from oil on gear and equipment. Sampling unoiled areas first, then lightly oiled areas and finally heavily oiled areas can minimize cross-contamination. Personal and SCUBA equipment should be exchanged or cleaned between sites if it becomes contaminated.

Study Design Implementation

- If recent SCAT data are available, it may not be necessary to conduct and overflight or ground survey. Otherwise, if practical, conduct an overflight of the entire affected area to observe the extent of visible contamination over known kelp-boulder fields.
- If an aerial survey is not feasible, survey from boats. Use topographic maps, nautical charts, vertical aerial photographs, or other detailed maps to record observations. Set a GPS in track mode and take a photograph of the date/time screen so photographs of surface kelp, if any, can be geo-referenced later. Observations should include:
  - Locations and approximate lengths of kelp-boulder fields in the oiled area. This will only be feasible when surface kelp is present
  - Approximate lengths and degree of oiling of the shoreline adjacent to kelp fields (if applicable)
  - Locations of access points, major landmarks, and potential ground-truth and reference stations

Note: Overflights may not be adequate enough to locate and identify kelp fields. For example, overflights are not likely to succeed in the Beaufort as there is no surface forming kelp, but could be useful in shallow areas (e.g., barren grounds of the Aleutians). It is highly advisable to consult with local experts and to select sampling sites based on trajectory modeling or similar.

- If available, boats rigged with high definition sonar equipment would facilitate the location of kelp-boulder fields, particularly in areas with low visibility, areas where surface kelp is not present, or under other conditions limiting the identification of kelp fields.
- Based on the study design and/or sampling strategy outlined before going into the field, establish a minimum of three (preferably five) transects spaced at least 30 m apart (recommended). When sampling kelp fields, transects should encompass the entire width of the kelp field.
- When establishing transects:
  - Record the transect location using a GPS and accurately plot the transect location on a map or aerial photograph
  - If possible, permanently mark the transect location using "front" and "back" flexible markers attached to the bottom/substrate that line up with the transect. Markers will need to be anchored. Consider placement carefully to minimize loss due to vandalism, erosion, ice scouring during winter, etc. Place a leaded line or stainless wire between the stakes so that they can both be relocated
  - Record the transect angle with a compass so it can be re-surveyed at a later date, even if one flexible marker is lost; note whether the angle reading is magnetic or corrected to true north
- For each transect, record:
  - Date, time, weather conditions (e.g., wind direction and speed), water clarity
  - Length of the transect (in meters) and of the sampling zone
  - General characteristics of the kelp field condition including death or discoloration
  - Oiling type description, thickness, and % cover on the substrate and vegetation
  - Sediment type (grain size)
  - Presence, condition, and /or altered behavior of visible biota such as amphipods, gastropods, crabs
- Divide each transects into sampling intervals of equal width based on the total width of the kelp field.
- Collect samples from each site (described below) and record the distance along the transect and GPS coordinates of each sampling site.
- Take pictures of the transect before and after sampling and pictures of each sampling site.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix B. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site using underwater cameras prior to sample collection to document the station conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling station (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If time allows, a calibration exercise is needed to ensure that all field teams consistently perform kelp-boulder field sampling.
- Decide if the site meets the selection criteria in terms of degree of oil exposure, physical setting, accessibility, and absence of other sources of contamination or degradation.

- Prioritize sample collection from each site based on logistics, sample storage, etc. as follows (in order of importance):
  - Collection of sediment, water, source oil, or other types of non-specific kelp habitat samples (see appropriate guidelines)
  - Estimation of percent vegetation cover and kelp oiling
  - Collection of infauna, fish and invertebrates, and tissue samples
  - Collection of kelp specimens
- At each sampling site, measure recommended physical/chemical parameters: salinity, water and air temperature, dissolved oxygen, turbidity, PAR, and irradiance. Follow instrument manual descriptions.
- When establishing quadrats at a site along a transect:
  - Take underwater photographs or video of each entire quadrat from an angle as vertical as practical. All photographs or videotape should be described in the photo log, located on a simple sketch map of the site, and noted on the data sheet. Photographs or videotapes should include:
    - The general station location and setting, showing permanent stakes
    - Examples of kelp field distribution and condition along the transect
    - Sites where samples were collected
    - Representative examples of the extent and degree of oiling
    - Examples of services provided by kelp-boulder field habitats (animal use, etc.)
  - If possible, describe the sediment composition and sediment oiling
  - Prior to sampling, note and describe any associated biota, including their behavior
  - For detailed sampling to characterize impacts on kelp-boulder fields within at least 3-5 (6 to 10 preferred) randomly placed 1 m^2 quadrats per transect at the kelp-substrate interface around the holdfast, and characterize the following:
    - Visual estimation of live and dead kelp coverage, including live and dead percent cover
    - Visual estimation of other sessile organisms including macroalgae, coralline algae, sponges, bryozoans, etc. ***Note:*** This will require technical knowledge for accurate species identification
    - Count mobile organisms greater than 2 cm
    - Visual estimation of kelp impact index:
      - No oiling - kelp appears to look natural, no evidence of oiling
      - Trace impact - kelp shows minor necrotic lesions
      - Light impact - kelp shows considerable necrotic lesions on <50% of leaves
      - Medium impact - kelp shows necrotic lesions on >50% of leaves
      - Heavy impact - kelp dead

      Note: Similar impact indices could be used for sessile species (e.g., sponges and coralline algae) as these organisms are important to boring species, which are in turn important to fish.
  - As needed, harvest at least 5 randomly selected plants with above structures (holdfast, stype, and blade) intact for laboratory measurement of biomass and age. Cut each plant at its base from the substratum with a knife. Samples can be stored whole. Place harvested material into high strength plastic bags, appropriately labeled and tied shut. Keep cold (~4° C) until processed
    Note: SCUBA will be required to sample the kelp fields.
- If faunal organisms are observed, or if key species are expected to occur (ecologically important prey species, etc.), sampling can be conducted to document the presence, composition, and general abundance of organisms. An experienced kelp-boulder field ecologist and statistician should be consulted to plan and carry out quantitative studies. For quantitative surveys of selected shoreline transects or habitat areas:
  - Take photographs with an underwater camera and record the GPS coordinates of each collection site

- For species presence/absence, composition, or density estimates use quadrats (0.25 $m^{2,}$ or larger if appropriate). Survey randomly 3 quadrats within sampling intervals of the random transects. Perform survey from at least 3-5 transects. If the degree of oiling is variable within the kelp field, perform surveys at 3-5 unoiled/least oiled transects and 3-5 oiled transects
- Vacuum all material within the quadrat at the kelp-substrate interface around the holdfast to a sediment depth of 10 cm using an airlift sampler:
  - Attach hoses and airlift sampler to the compressed gas tank equipped with a regulator
  - With the regulator closed, open the main tank valve all the way and turn the valve back one full turn
  - Slowly open the regulator until the pressure in the range of 20-45 psi (140-310 kPa)
  - Place the inlet of the airlift sampler on the quadrat substrate
  - Deliver short pulses of compressed gas to the sampler to suction the contents of the quadrat and approximately the top 5 cm of substrate
  - Filter contents through a 250 μm sieve and transferred into sampling jars
  - Preserve samples in 10% formaldehyde buffer (in sea water) or 95% ethanol (less ideal) until processed in the laboratory, and stain (such as Rose Bengal) if appropriate
  - Place a waterproof label with the station location, sample number, and date on the sample container

  Note: Modifications to this method may be needed when sampling the kelp-substrate interface on boulders.
- If possible and visibility allows, photo-document the sampling locations, sample collection methods, organisms observed, any obvious oil impacts, etc. and keep a good record of the quadrat-photo sequence

- If tissue analyses are planned for larger infauna, such as bivalves, follow the Shellfish Tissue guideline. Briefly:
  - Wrap each individual specimen in pre-cleaned aluminum foil, and freeze the sample as soon as practical
  - Take care to avoid cross contamination during sampling and handling
  - Clean sampling equipment between collections
  - Ship biological and sediment samples on ice overnight to the laboratory conducting the analyses
- If direct surveys (via SCUBA) are unpractical or unsafe, consider using high-definition underwater videography deployed from vessels to document vegetation condition and fauna presence. This type of sampling requires previous training. Briefly:
  - Mount the underwater camera in a 'down-looking' orientation on a towfish deployed directly off the stern of the vessel
  - Allow the camera to follow the kelp field contour above the canopy
  - Maintain the field of view as constant as possible (1 m^2)
  - The vessel speed should be held as constant as possible (about 1 m/s) to facilitate estimation of distances
  - Conduct straight line underwater video transects (randomly selected) perpendicular to the shoreline and encompassing the width of the kelp field
  - Carefully catalogue all underwater videos and ship to the appropriate laboratory for processing and interpretation. Post-processing of underwater videos can be used to estimate kelp vegetation coverage and condition
- Take sediment samples and samples of representative invertebrate species for chemical analysis particularly from areas where kelp-boulder field impacts are visibly obvious. If oiled areas are sampled, fauna and sediments from reference areas should also be collected (see Shellfish Tissue and Subtidal Sediment guidelines).

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Complete the Chain of Custody form, noting where each sample was collected, sampling device used, time/date of collection, size and container type, and sampler name (See Chain of Custody guidelines).
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the following on the field sample form for each sample:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample #, date/time, water depth
  - Describe the oiling conditions (using standard shoreline assessment terminology) (if applicable), weather conditions (e.g., wind direction and speed), water clarity
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Tape lids on sample bottles so that they do not accidentally come off.
- Ship known oil-contaminated samples separate from non-contaminated or low-contaminated samples to reduce risk of cross-contamination.
- Ship any samples preserved in formalin or other chemicals as hazardous goods. Include Chain of Custody forms.
- Refrigeration temperature shall be recorded upon sample storage, and monitored and recorded periodically to ensure proper refrigeration.
- Use packing material, such as bubble wrap, around containers to prevent breakage during handling and shipping.
- Ship samples directly to the laboratory as soon as possible with complete chain of custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended storage temperatures.

Analytical Methods

- Refer to those under Shellfish Tissue and Subtidal Sediment guidelines.

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Appendix A
General Guideline for SCUBA Sampling of Kelp Fields

The guideline below provides a series of recommendations to ensure the safety of underwater sampling. These recommendations are not intended to be comprehensive. Consult additional sources of information on underwater safety. Remember that the success of underwater operations result from careful planning and observation of safety precautions.

- If underwater hazards are identified (e.g., strong currents, unsafe wave conditions), cancel or suspend any underwater sampling until the hazard is no longer an issue.
- SCUBA is not recommended when oil slicks are present at the water surface; however, it may be acceptable if only light sheens are present.
- SCUBA activities are NOT to interfere with response operations
- Night SCUBA diving is not recommended.
- When planning underwater activities follow these recommendations:
  - Have all emergency information and phone numbers handy
  - Inspect all essential underwater equipment prior to initiation of underwater activities
  - Establish a communication system between underwater samplers, and between underwater samplers and personnel at the surface
  - Document all planned depths and sampling type
  - Be aware of your surroundings and consider environmental conditions, currents, tides, visibility, air/water temperature, accessibility, and other natural and man-made hazards
  - Be aware of other activities in the area that may interfere with the dive or that pose a safety hazard (i.e., vessel traffic, noise, pollution, etc.)
  - Thermal protection should be ensure at all times by having wet or dry suits with hoods available. Dry suits are recommended for sampling in waters below 50°F, or when sheen is observed at the water surface
  - Remember that because cold affects dexterity, underwater activities in cold waters must be shorter than those in warmer waters
  - Sampling team members should be trained in cardiopulmonary resuscitation (CPR) and first aid, and should be able to recognize and treat any signs of hypothermia
- Specifically for SCUBA:
  - All divers must be properly trained on safe underwater procedures
  - Ensure that there are enough air tanks available for the duration of sampling activities
  - Maintain a Dive Log
  - SCUBA requires a 2-buddy system and should remain in visual sight of each other during ingress/egress and while underwater
  - A warning flag must be displayed at the dive location
  - Document and report any incidents that occurred during underwater sampling

Appendix B
Supporting Documentation - Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on waterproof paper. Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink. Notes taken under water may need to be transcribed onto field data forms.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:

- Kelp Field Data Form – 2 parts: General information, Transect data

Kelp-Boulder Field Data Sheet - General information (one per field)

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder/ Affiliation: _____

3 Other team members / Affiliation: _____

4 **Site Descriptors**

Site Name/ID: _____

Latitude: _____ Longitude: _____

Habitat setting (check one): _____ _____ Subtidal - Depth (m): _____

Field Size: Width (m) _____ Length (m) _____

Overall field condition: _____

Kelp percent oil coverage (%): _____

5 **Physical/Chemical Parameters**

Bottom salinity (ppt): _____ Air temperature (C): _____

Bottom temperature (C): _____ Bottom dissolved oxygen (mg/L): _____

Weather/Cloud cover: _____

Wave height (m): _____ Secchi depth (cm): _____

PAR ($uEm^{-2}s^{-1}$): _____ Oiled Condition (check 1): _____ None

_____ Sheen

_____ Light

_____ Moderate

_____ Heavy

	Irradiance	Depth (m)
Value 1		
Value 2		
Value 3		

6 **Sketch**

Drawing Legend	
~~	Water line
▬▬▬	Kelp field
----------	Transects

Additional information:

7 Photos:

Number	Description	Number	Description

Notes

Kelp-Boulder Field Data Sheet - Transect data (one per transect) **Page 1 of 3**

1 Date: _____ Time: _____ Site Id: _____ Team ID: _____

2 Data Recorder / Affiliation: _____

3 Other team members / Affiliation: _____

4 Transect ID: Transect Length (m):

	Waypoint #:	Latitude (DD)	Longitude (DD)	Transect Angle
Front Stake				
Back Stake				

Photos:

Number	Description	Number	Description

5 Sketch

Drawing Legend	
FS	Front Stake
BS	Back Stake
~~	Water line
▬▬▬	Kelp field
----------	Transect
□	Quadrat
V	Plant Sample
S	Sediment Sample
W	Water Sample
I	Invert. Sample

6 Transect Characterization

Interval Length (m)	Kelp cover (%)	Kelp condition (e.g., chlorosis, necrosis)	Vertical oil interval	Oil Cover (%)		Sediment type (grain size)
				Kelp	Substrate	

Notes:

7 Quadrat Characterization **Page 2 of 3**

Quadrat Location	Dist. from transect (m)	Cover (%)		Sample taken (yes/no)	Kelp species and associated species (e.g., macroalgae, sessile and mobile invertebrates)	Count, % cover (if applicable)	Sample taken (yes/no)
		Live	Dead				

[1] **Kelp Oiling impact scale:**
1 - None - kelp appears to look natural
2 - Trace - kelp shows minor necrotic lesions
3 - Light - necrotic lesions on <50% of fronds
4 - Medium - necrotic lesions on >50% of fronds
5 - Heavy - kelp dead

8 Point Sample Collection (use appropriate field data sheets for point samples)

Vegetation samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Vegetation samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Sediment samples for grain size analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Water samples for chemical analysis

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Invertebrate samples for biological analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Invertebrate samples for chemical analyses

Sample ID	Latitude (DD)	Longitude (DD)	Notes

Other samples

Sample ID	Latitude (DD)	Longitude (DD)	Notes

EPHEMERAL DATA COLLECTION GUIDELINES:

OTHER AQUATIC RESOURCES

Guidelines for Collecting Ephemeral Data in the Arctic: PLANKTON

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting plankton samples during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment exposure and injury evaluations.

Sampling Objectives

Study exposure

- Assess the risk to plankton from exposures to oil constituents
- Quantify the composition, distribution, biomass, and densities of plankton (including ichthyoplankton, zooplankton, and phytoplankton) in background and oiled nearshore waters

Quality assurance/quality control

- Ensure the integrity the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of biological characterizations

Collaboration

- Support other ongoing efforts including, but not limited to modeling of impacts to water-column resources (see Water, Shellfish Tissue, Fish guidelines), toxicity testing for injury assessment and assessing injury to higher trophic organisms that prey on plankton.

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Plankton are difficult to sample because of their inherent heterogeneity of distribution over space, depth, and time.
- The following terminology is used to define general to specific sampling geographies:
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Site = a specific point at which samples are collected or observations are made

- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with plankton sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.
- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their recommended holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours, if possible. However, working from a vessel with deck lights that allow safe operations would permit nighttime sampling. This guideline may not apply during winter or much of the fall.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled by the spill and unoiled reference areas.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what areas have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be affected by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize sampling locations. In this case, highest priority samples are to be collected from nearshore water adjacent to sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiling nearshore water adjacent to sensitive/productive areas that are likely to be oiled by the spill in the near future is also a priority. Sampling at unoiled "control" areas and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Plankton samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter. Sampling frequency should be defined in the study design.
- Use a computer or conceptual model of the extent of water-column contamination or an appropriate power analysis to determine the number and location of samples:
  - Sample along exposure gradients, starting in the cleanest zone, at regular intervals proportional to the exposure area
  - Minimum guidelines under normal conditions are at least three samples per waterbody location of relatively uniform oceanographic and oiling exposure
  - If logistical limitations are a concern, sample collection MUST be prioritized by selecting a minimum of 1 reference/pre-oiled location and 2 heavily oiled locations
- When appropriate take duplicate samples from the same site and following the same steps as the preceding sample. This is not the same as collecting three replicates from each site/depth. A minimum of one duplicate sample should be collected at every third sampling site, or following specifications of the work plan. As a general rule of thumb, duplicates are 10% of all samples, but consideration should be given to sample storage capacity. Do not split samples unless these types of samples are specified in the work plan.
- When present, plankton samples from nearshore lagoons with connectivity or potential connectivity to the marine environment should be obtained.
- Preferably, plankton samples should be collected from boats, though collection by wading in shallow water is also possible.

Collaboration

- Plankton samples from nearshore areas can be collected in conjunction with water, snow, or ice sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers

Note: The amount of equipment required depends on the sampling plan, desired sample volumes, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.

- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)

- Conical nets: 10-µm Nitex® mesh or similar net (for larger phytoplankton), tow-net (mesh sizes range from 64 µm to 505 µm; e.g., bongo net) ideally equipped with flowmeters – for collection of plankton samples.
- Zooplankton bucket
- Niskin or similar sampler
- Shadowed Image Particle Profiling and Evaluation Recorder (SIPPER) (optional) – Though expensive and requiring significant logistics for shipping and deployment, use of this equipment avoids the issue of contamination of plankton nets and provides in-situ information
- Pencils, waterproof pens, waterproof labels, markers
- Ice corer for sampling under ice
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves preferred for water sampling under extreme cold conditions
- 1 L glass jars, amber glass preferred – for plankton and water sampling
- 100 mL plastic centrifuge tubes – for samples collected for purposes other than chemical analysis
- Lugol and 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal) – for sample preservation
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- Field notebook (waterproof paper), other guidelines as needed (Water, Fish, Subtidal and Intertidal Sediment guidelines)
- GPS, camera (with spare batteries), and photo scales
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment are not available):
- Sufficient quantities of pre-cleaned or disposable single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control
- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination
- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.

- Check for major rips or holes in the mesh, especially in the lower 1/3 of the net. If holes are detected, repair them or replace the net.
- Make sure that there are no bubbles in the flowmeters. Check to insure that the flowmeter rotor spins freely and does not wobble, i.e., the shaft is not bent.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.
- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples collected for chemical analysis. To decontaminate sampling nets if they become visibly oiled and any other sampling equipment prior to each use:
  - Wash nets/equipment/sampling utensil with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, “background” water from an up-current clean area can be used.
- Take precautions to avoid cross-contamination of the site from oil on boots and other gear.
- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts, and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.
- Contamination by surface slicks is of great concern. Document presence of slicks, weather, wave conditions, etc. which might suggest mixing of surface oil during sampling.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- Avoid sampling under sheens and oil slicks, but if unavoidable, clear surface oil prior to plankton sampling. If possible, place sorbent boom upstream of the sampling site to temporarily divert oil. Alternatively, use sorbents to remove oil inside the net prior to collection of plankton.
- If possible consult a plankton expert when developing the study design to determine what net size is appropriate for the study. Modifications of this guideline may be needed depending on the types of plankton that are being targeted and/or the nets that are available for plankton sampling. Note that species composition, plankton size, life stages, etc., captured will vary depending on the net mesh size used for sampling. For example:
  - Mesh size as small as 64 µm can be used to sample phytoplankton and micro-zooplankton

  - Mesh size up to 3 mm can be used to capture the largest zooplankton
  - Standard mesh size used in the Arctic are typically 150 µm and 505 µm for small and large taxa, respectively
- Vertical, double-oblique and horizontal deployment speed needs to be considered and recorded for each type of net used in plankton tows.
- To sample phytoplankton qualitatively (presence/absence, taxonomic surveys) (less ideal):
  - Lower a 10-µm Nitex® mesh or similar net to a given depth, allow it to settle for 30 seconds and pull it slowly to the surface
  - Slowly pull the neck back to the surface. If bow waves or obvious disturbances of the water column or water surface are observed discard sample and start again
  - After retrieving the net from the water, place the mouth of the net into a 1 L sample-collecting bottle and drain the sample. Collect samples in triplicate
- To sample phytoplankton quantitatively:
  - Open the, Niskin or similar sampler (e.g., 4 L) by raising the end seals and set the trigger mechanism
  - Lower the sampler to the desired depth, send the messenger down to close the seals, and retrieve the sampler to the surface
  - Carefully, remove the sampling bottle, gently shake it and transfer contents into a 1 L sample bottle leaving a 3-5 cm headspace
  - Preserve samples with 3 mL of Lugol's solution or 0.05-1% by volume, followed by preservation with 20 mL of an acidified formalin solution (2% by volume). The second preservation step is important for preserving the color of algae. If space in coolers is limited and/or there are concerns about transport of preservatives, omit the second preservation step
    - Lugol's solution can be prepared as follows: 100 g I, 200 g KI, 200 mL glacial acetic acid, and 2000 mL of distilled water. This solution MUST be stored in a dark bottle because of its sensitivity to light
    - Acidified formalin solution can be prepared as follows: equal volumes of formaldehyde (37%) and glacial acetic acid
    - Both of these solutions can be prepared in advance and used up to 2 months of preparation. Label each bottle including the name of the preservative and the date of preparation
    - Cap the bottle and place it a cooler, minimizing exposure to light
- To sample zooplankton quantitatively:
  - Select the appropriate zooplankton mesh size tow-net (64 µm to 505 µm) equipped with a flowmeter. A flowmeter is important because it allows an estimation of the volume of water that passes through the net. This information, in combination with counts of organisms caught in the net, zooplankton concentration per volume of seawater can be inferred
  - Soak the body of the tow-net in water for 2 minutes and rinse the net to dislodge any attached material
  - Attach the zooplankton bucket/bongo with plug in place, and fill a Nalgene squirt bottle with seawater that has been filtered through the net mesh
  - For vertical sampling, which targets smaller zooplankton, lower the net to the appropriate depth in a vertical position, allow the net to settle for 10 seconds, and raise it vertically at average speed of 1 knot (0.5 m/s) to minimize avoidance of the net by fast-swimming zooplankton. This sampling should performed from a stationary or anchored ship, or from a fixed platform
  - For double-oblique sampling, which targets larger and more mobile zooplankton, lower the net wide from the surface to the desire depth, continue sampling at the desire depth for 30 seconds, and retrieve the net back to the surface. This sampling should performed with the ship moving at an average speed of 2 knots (~1 m/sec)
  - For horizontal sampling, useful when characterizing zooplankton at different depths, lower the net to the target depth, pull the net at that depth for approximately 5 min at average speed of 1

knot (0.5 m/s), and quickly retrieve the net back to the surface. This type of sampling is also useful for surface sampling in shallow waters. In these cases, place the net on the water surface and once it is half way submerged, start pulling the net at an upward angle
  - At the surface, rinse down the outer sides of the net 3 times with seawater avoiding the net opening, and do not let the net drop below the surface
  - Separate the bucket from the net, place the lower end of the bucket into a sample bottle. Remove the plug and drain the bucket contents
  - Rinse the bucket contents into the sample tube with the squeeze bottle previously filled with filtered net water
  - Preserve zooplankton samples in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal)
  - Do not freeze preserved samples
- Rinse the net and bucket with clean seawater between sites.
- When sampling through ice:
  - Clear loose ice and snow away from the sampling location and drill through the ice
  - Clean the drill-hole area from potential sources of contamination, and allow several minutes for the water to flow freely under the ice before taking a sample
  - Carefully lower the net and follow the rest of the sampling procedure and preservation as described above
  - To sample ice plankton, collect a 7.5-10.5 cm diameter core and cut the bottom 2-4 cm of ice. Melt the ice in surface water filtered through 0.2 µm polycarbonate membranes, recording the volume of water used
  - Preserve samples for phytoplankton analysis with Lugol's solution followed by an acidified formalin solution (see Ice guidelines)
  - Preserve samples for zooplankton analysis in 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal)
- To sample plankton quantitatively *in-situ* (only if resources and personnel are available): The SIPPER (Shadowed Image Particle Profiling and Evaluation Recorder) can be used to collect high-resolution information on the distribution of zooplankton, phytoplankton, larval fish, and detritus within a 100 cm^2 sampling area as it moves through the water. The SIPPER is also equipped with a CTD (conductivity, temperature, depth) scan, oxygen sensor, fluorometer, transmissometer and CDOM (colored dissolved organic matter) fluorometer. When using this sampling strategy:
  - Tow the SIPPER horizontally and vertically through the water at speeds between 1-4 knots. SIPPER tows require at least an hour per 100 m depth
  - Conduct multiple tows at day and night in control/least oiled and contaminated waters
  - Sampling locations, depths, number of transects, and aerial extent, will be adaptively selected. Ideally, distinct areas of relative uniform exposure would be sampled once (1 SIPPPER vertical profile) every 100 m
  - SIPPER operation will require expertise and training, and will require on board ship space
- Plankton samples from nearshore areas should be collected in conjunction with water sampling for basic environmental parameters (i.e., temperature, salinity). If practical, deploy a CTD to collect oceanographic parameters at every sampling site.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each plankton sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.

- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample # on both the label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (plankton)
  - Sample #, date/time
  - Sampling method (bottle, net), sample collection depth, distance from shoreline
  - Sample #; date/time; station location; GPS coordinates, water depth
  - Characteristics of suspended material in the water sample: texture, color, biota, vegetation, debris, odor, etc.
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.
- If sample volume is split between two jars, both jars should receive the same sample ID and be recorded on a single line of the Chain of Custody form.
- Documenting oil exposure is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Preservation/Holding Times

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oiled samples separate from lightly or unoiled samples to reduce risk of cross-contamination.
- Immediately following collection, place all preserved plankton samples in a cooler. DO NOT FREEZE samples preserved in formalin. Refrigeration temperature shall be recorded upon sample storage and monitored and recorded periodically to ensure proper refrigeration.
- In below-freezing temperatures, collapsible jugs of warm water can be used in the cooler between samples to prevent them from freezing.
- Preserve samples immediately after collection, and discard samples not preserved within one hour of collection.
- Do NOT use freshwater when sampling and preserving plankton samples.
- Do NOT keep plankton tows that contain sediments.
- Tape lids on sample bottles so that they do not accidentally come off.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical, overnight (preferred), with completed Chain of Custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Key References

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Appendix A
Supporting Documentation- Field Data Form Examples

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached form:
- Plankton Sample Collection Form

NRDA Sample Collection Form - PLANKTON

Lead Sampler's Name/Phone		**Sampler Team Code**		**Vessel Name**	
Lead Sampler's Affiliation		**Resource Group**		**Wind Speed**	
NRDA Contact/Phone		**Resource Group Leader**		**Wind Direction**	
Incident Name		**Habitat (e.g., lagoon)**			
General Location Description		**Sample date (mm/dd/yyyy)**			

Location Code	Sample Number	Sampling Gear / Size	Tow Start Time	Tow Start Latitude	Tow Start Longitude	Flow Meter Start	Tow End Time	Tow End Latitude	Tow End Longitude	Flow Meter End	Tow Depth or Max Depth	Tow Length	Sample Container	Sample preservation	Instrument Data	Sample Notes
NRDA Sample Grid ID	*TEAM ID - Sequential Numbers*	*Net type / Mesh Size (uM)*	*(24-hr clock, local time)*	*Latitude in DD XX.XXXX XX*	*Longitude in DD YY'Y.YYY YYY*		*(24-hr clock, local time)*	*Latitude in DD XX.XXXX XX*	*Longitude in DD YY'Y.YYY YYY*		*Depth (m)*	*Distance (m)*	*Jar, tube, etc.*	*Preservative (Formalin, Ethanol, etc.) or NA*	*Instrument Used / Metadada File Name*	*Indicate if noted below*

Survey Notes - (flow meter serial number, weather, wildlife observed, photos, etc.)

Relinquished by:				**Received by:**			
Date	**Time**	**Signature -Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**

Guidelines for Collecting Ephemeral Data in the Arctic: FISH

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on collecting nearshore, shallow-water fish, including spawning aggregations, juveniles, and adults, during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations. It is assumed that all sampling will be done from shore or using a skiff launched from shore due to limited logistics early in a spill. The major nearshore habitat (waters <10 m deep) types for fish in the Arctic are coastal beaches, lagoons, river deltas, estuarine, and open waters (waters 10-20 m deep). Key differences among these habitats are temperature, salinity, and wave exposure.

Sampling Objectives

Characterize oil

- Determine the concentration and composition of oil compounds in biological tissues compared to background concentrations

Study exposure and injury

- Document the fish species and life stages present and at risk of exposure to oil constituents in nearshore coastal waters, estuaries, and lagoons
- Quantify the composition, distribution, biomass, and densities of fish (spawning aggregations, eggs, larvae, juveniles, and adults) in background and oiled nearshore waters and lagoons
- Quantify oil chemicals, chemical metabolite and biomarkers of exposure in fish
- Document acute fish mortality in nearshore waters

Quality assurance/quality control

- Ensure the integrity the sample(s) throughout sampling, transport, and storage
- Ensure the reliability of biological characterizations

Collaboration

- Support other ongoing efforts including, but not limited to, modeling of oil transport, exposure and impacts to water-column (see Water, Snow, Shellfish Tissue, and Plankton guidelines), toxicity testing for injury assessment, and assessing the exposure and risk to higher trophic organisms consuming contaminated prey

Before Field Sampling

- Assure that all personnel have required safety training and protective equipment for Arctic field work (not described in this guideline).
- Arctic weather conditions (e.g., wind direction and speed) are variable within a short timeframe. Be prepared for changing weather conditions, be aware of your surroundings, and take precautions to ensure the safety of the sampling team.

- Special safety training and equipment may be required if small boats are used to sample fish.

Study design

- It is important to have a defined sampling strategy prior to conducting fieldwork. Fish are difficult to sample because of their inherent heterogeneity of distribution over space, depth, and time.
- Decide in advance if fish will be sampled to quantify biological/ecological parameters (e.g., presence/absence, relative abundance, life stage, size class, etc.), for chemical analysis, or both. Adapt the study design and guidelines accordingly.
- The following terminology is used to define general to specific sampling geographies
  - Area = general area of uniform characteristics, such as degree of oil exposure, physical setting, habitat types present, etc.
  - Location = a specific location that is representative of the area and contains the type of habitat to be sampled, such as an eelgrass bed or lagoon
  - Site = a specific point at which samples are collected or observations are made
- Plan ahead the number of locations and number of sites per location, taking into account level of effort, potential logistical limitations, weather conditions, and other issues that may compromise sample integrity.
- Review the guideline and resolve any area-specific issues. Area-specific modification of the guideline may be needed based on environmental conditions, geography, access to remote areas, and shipping capabilities.
- Some elements of the study design will be dependent on the availability of small boats for nearshore work and plans should be adjusted accordingly.
- Contact the laboratories that will be receiving field samples for analysis and assure that they have the capacity to receive and analyze samples from the study. Follow relevant guidelines from the laboratory and consult with them about necessary modifications.
- The sampling strategy should have flexibility to be adjusted based on conditions in the field.
- Consult appropriate guidelines for the collection of other environmental media and biota concurrent with fish sampling. Tarballs, sheens or other oil residues can be collected opportunistically for chemical analysis and fingerprinting.

Equipment

- Review the list of sampling equipment/containers, make adjustments as needed, and assure that all essential field materials are ready to be taken to the field.
- If not all sampling equipment is available, consult the alternative equipment guidelines or determine if other appropriate options are available.
- It may be necessary to coordinate with the laboratory that will receive the samples to assure that acceptable materials and conditions are used for sampling and sample storage and shipping.
- Do as much material preparation prior to field deployment, including: labeling sample jars using permanent markers or laboratory labels (e.g., peel and stick waterproof labels); pre-cleaning aluminum foil for sample storage, etc.
- Make sure that all essential equipment is in working order and operational under Arctic field conditions, and that spare equipment and/materials are available.
- Store solvents carefully to prevent spillage. Follow regulations regarding the shipment and storage of chemicals.

Sampling Areas and Timing

- Follow a sampling plan/work plan if one is available.

- If a sampling plan is not available for ephemeral data collection immediately after a spill, data collection should focus on collecting samples from a range of unoiled, likely to be oiled, and already oiled areas.
- It is important to obtain reliable data that account for spatial and temporal variations of oil impacts.
- When sampling in remote areas with limited shipping capabilities, plan ahead to make sure that the integrity of samples is not compromised by ensuring that the processing laboratory receives the samples within their holding time. Remember that it may take multiple days for shipments from remote areas to reach a laboratory facility. This last stage is the most important and requires due diligence until the samples are safely delivered.
- The number of locations and number of sites per location need to be considered accordingly, making sure that there is enough space in the coolers to accommodate all samples without sacrificing their integrity.
- Plan all sampling strategies within daylight hours; sampling in the dark, even with headlamps, is not recommended for shore-based activities. This guideline may not apply during winter or much of the fall. However, working from a vessel with deck lights that allow safe operations would permit nighttime sampling.
- The challenges of collecting samples in remote areas, particularly during winter, are great and require adequate planning and careful field implementation to attain the data quality required to meet the objectives of the sampling plan.

Area selection

- Sampling locations should be representative of areas that have been or may be oiled and unoiled reference locations.
- Sampling locations should be characterized by benthic habitat: soft bottom with eelgrass, cobble with understory kelp, steep bedrock outcrop, inundated tundra, unvegetated sand or gravel bottom, etc. Sampling areas may also be characterized as estuarine, lagoon or nearshore open water.
- Use satellite images, charts, maps, ShoreZone images, etc. to select sampling areas.
- When present, fish in nearshore lagoons with connectivity or potential connectivity to the marine environment should be sampled.
- Use trajectory models, conceptual models, overflight information, SCAT data, or other tools to determine what locations have been oiled and which ones are likely to be oiled.
- Samples should also be collected from locations known or suspected to be impacted by other natural or anthropogenic sources of contamination (e.g., oil seeps, coal, peat, mining, combustion engines), as these will be important to differentiate background sources and levels of contamination.
- It may be necessary to prioritize site selection. In this case, highest priority samples are to be collected from oiled nearshore/offshore areas that are sensitive habitats, biologically productive, or highly relevant for human use. Collecting pre-oiled fish from sensitive/productive sites that are likely to be oiled is also a priority. Sampling at unoiled “control” locations and sampling other sources of contamination should be prioritized based on the ephemerality of the data and relative importance to developing a NRDA case.
- Depending on the study design and sample storage space limitations, it may not always be necessary to collect samples to bring back to the laboratory. Some parameters of interest, including fish species identification, fish kills, relative abundance, life stage, size, etc. can be documented in the field. If space is limited, prioritize obtaining and storing samples for chemical analysis while recording other data in the field.
- On the other hand, in severe weather conditions, it may be preferable to collect samples and transport them to the laboratory for identification, counting and other processing.
- Fish samples should be collected pre-oiling, if possible, as soon as practical after oiling, and periodically thereafter.

- Use a computer or conceptual model of the extent of water-column contamination or an appropriate power analysis to determine the number and location of samples.
  - Minimum guidelines are at least three beach seines, small midwater trawls, or bongo tows per area of relatively uniform exposure or distinct waterbody, performing two daytime beach seines per station (preferred). While two additional nighttime beach seines can be performed, special arrangements are needed to ensure safe nighttime sampling
  - If logistical limitations are a concern, prioritize sample collection by selecting a minimum of one unoiled/pre-oiled location and two oiled locations
- Sample along exposure gradients, starting in the cleanest location and at regular intervals proportional to the exposure area.
- Fish can be sampled from boats, the shoreline, or by wading in shallow water.

Collaboration
- Fish samples can be collected in conjunction with water, snow, ice, shellfish, plankton, and sediment sampling.
- Close collaboration and coordination with other ongoing ephemeral sampling efforts is important.

Field Sampling Methods

Sampling Equipment/Containers
Note: The amount of equipment required depends on the study plan, desired sampling capacity, and logistics. Analytical laboratories may provide required sampling and sample storage and transport materials – contact the receiving lab before preparing to collect samples in the field.
- Boat with motor (if available)
- Coolers – for sample storage and transport
- Ice packs/Collapsible jugs − for storage temperature regulation (if ambient temperature exceeds 4°C)
- Thermometer or temperature logger (1 per cooler)
- Nets:
  - Short-handled dip net − for handling live fish, one per field team, but have extras in case of net damage or loss
  - Plankton nets such as a bongo net − for egg, larvae, and small juvenile fish sampling; may be used under ice
  - Vertical nets such as a beach seine or gillnet − for nearshore sampling of larvae, juvenile, and adult fish
  - Trawl nets such as a midwater or bottom trawl − for nearshore sampling of larvae, juvenile, and adult fish
  - Beam trawls − for nearshore sampling of epibenthic fish
  - Fixed and drift nets such as a fyke net − for shallow lagoons and/or nearshore sampling of juvenile and adult fish
  - Fish traps − for nearshore sampling of adult forage fish; may be used under ice
  - Other nets or traps may be appropriate and could be used as available
- Large tub or buckets (several)
- Forceps for moving small fish
- Battery powered aerators − for maintain fish alive
- Fish club- for sacrificing fish
- For field data collection: sampling trays, hand counter, meter measuring board with 1 mm divisions − for measuring fish, two per field team; portable electronic balance and calibration standard − for fish wet weight, one per field team
- Dissection kit with scalpel, scissors, and forceps − for collection bile and tissue samples, plus additional scalpel blades (enough to change between each sample)

- Pencils, waterproof pens, waterproof labels, markers
- Ice corer for sampling under ice
- Disposable nitrile gloves (preferred), insulated nitrile-coated gloves (less ideal)
- Insulated shoulder-length rubberized gloves (preferred) for water sampling under extreme cold conditions
- Sampling jars – certified organic-clean glass jars (solvent rinsed) with Teflon-lined lids and labels:
  - 4 mL amber glass vials (for fish bile samples) with cloth labels
  - 4 oz. amber glass jars for tissue samples
- Plastic vials (for fish and tissue samples not used for chemical analysis)
- 10% buffered formalin (in seawater) (preferred), or in 95% ethanol (less ideal)– for sample preservation
- Pre-cleaned aluminum foil
- Ziploc bags − for storing fish
- Sorbent pads
- Field Sample Forms (template in Appendix A)
- Chain of Custody forms (see Chain of Custody guideline)
- Evidence tape (see Chain of Custody guidelines)
- Fish identification field guides/charts
- Field notebook (waterproof paper), other guidelines as needed (Water, Snow, Ice, Plankton, Shellfish Tissue, Subtidal, and Intertidal Sediment guidelines)
- GPS, digital camera (with spare batteries), photo scales
- Packaging materials for glass jars (e.g., bubble wrap, sorbent pads, tape) – may be provided by the analytic laboratory
- Suitable disposal bags for oiled PPE and disposable sampling materials

Optional (if single-use sampling equipment are not available):

- Sufficient quantities of pre-cleaned or disposable equipment, single-use equipment are preferable. If equipment will be reused in the field, decontamination is necessary and will require the following materials:
  - Reusable sampling equipment
  - Laboratory-grade detergent (Liquinox or similar)
  - Solvents for cleaning sampling equipment – acetone, methanol, or hexane (Capillary GC Pesticide Residue Grade or equivalent) – consider shipping/airline regulations for solvents
  - Teflon solvent squirt bottles
  - Laboratory-grade, certified-clean distilled water (preferred), store-bought distilled water (less ideal); laboratory-grade detergent
  - Approved, sealed container for collecting solvent rinsate for disposal

Quality Assurance/Control

- Rinsate blanks should be collected if there is a risk of cross contamination from reuse of sampling equipment. After cleaning the equipment in accordance with the procedures described in this method, rinse the clean equipment with solvent or cleaning solution and collect the rinsate in a sample jar. Note on the field sample form where and how rinsate blanks were collected.

Good Sampling Practices and Decontamination

- Good field practices and the development of a consistent sampling routine will help provide for the integrity of the samples and their validity in environmental assessments.
- Disposable nitrile gloves should be worn when sampling and changed between each sample collected or as necessary to prevent cross contamination.

- Disposable nitrile gloves can be worn over low-profile insulated gloves (e.g., neoprene gloves) in cold conditions and should be changed between samples to prevent cross contamination if they become contaminated or damaged. If nitrile gloves are not available or will not fit over insulated gloves in cold conditions, insulated nitrile-coated gloves may be an alternative, but extra precautions will have to be taken to prevent sample contamination; gloves will need to be cleaned with soap and clean water between samples and should not come in contact with the sample or with the surfaces of glassware or tools that will be in direct contact with the sample. Similar precautions should be taken when using insulated shoulder-length rubberized gloves.
- To reduce the need for field decontamination, use pre-cleaned and/or disposable equipment and tools.
- If disposable sampling equipment are not available, reusable sampling equipment MUST be decontaminated between samples. To decontaminate sampling nets and any other sampling equipment prior to each use:
  - Wash nets and other large equipment with detergent and rinse with water. If detergent is not available, wash nets thoroughly with clean water
  - Wash sampling equipment with laboratory-grade detergent and clean with a triple clean-water rinse. Cleaning with laboratory-grade water is preferred, though store-bought distilled water is a less ideal alternative and, as a last resort, "background" water from an up-current clean area can be used
  - Rinse sampling equipment with methanol or acetone, followed by hexane (Capillary GC Pesticide Residue Grade or equivalent). Collect solvent rinsate for proper disposal or shipment to the lab as a rinsate blank. Allow solvents to evaporate from equipment before use. Do not work with solvents downwind of exhaust or other airborne hydrocarbon source. If solvents are not available, use a diluted detergent solution and fresh water, followed by a distilled water rinse. If transporting solvents is not feasible, use single-use sampling material
- Take precautions to avoid cross-contamination from oil on boots and other gear.
- Potential sources of contamination while sampling from vessels (exhaust fumes, oily surfaces) are a concern. Work up-wind of any exhausts, and designate clean areas for handling samples. Segregate dirty/clean areas. Layout clean surfaces to work on and replace frequently.

Sample Collection Methods

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Because GPS units will be used to record locations and times, make sure that all units are using the same coordinate system, datum, reporting units, and correct time. Follow the recommended GPS datum of the study plan, if one is available. Alternatively, set the default to WGS84.
- Record GPS coordinates for each sample site.
- Photograph the sampling site prior to sample collection to document the site conditions, as well as the sample collected. Make sure each photograph or series can be later associated with the corresponding sampling locations (e.g., through use of GPS Photo link software or by keeping a detailed photo log with waypoints and/or lat/long). Do not delete or alter any photographs. The numbering sequence of photographs uploaded from your camera must not have any gaps (see Field Photography guideline).
- If possible, avoid sampling under oil sheens and slicks, but if unavoidable, clear surface oil prior to fish sampling. If possible, place sorbent boom upstream of the sampling area to temporarily divert oil. Alternatively, use sorbents to remove oil inside the seine prior to collection of fish.
- For tissue sample for chemical analysis collect live animals. If live animals are not available, collect dead animals only if tissues appear to be fresh. Note the collection of live/dead animals on the field sample forms.
- If fish kills are observed, note the location, species, life stage, visible oiling and approximate number of fish observed dead in the water or along the shoreline. Take pictures of any fish kills and note

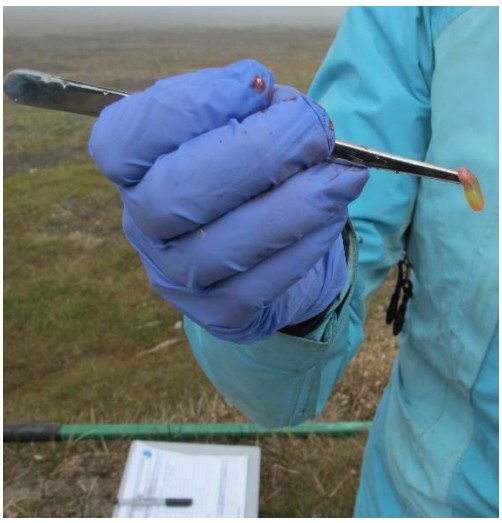
observations or evidence of scavenging. Consider collecting samples for to determine cause of death or for chemical analysis.

- Sampling modifications of this guideline may be needed depending on the types of nets available for fish sampling. Note that species composition, fish size, life stages, etc. will vary depending on the net use for sampling (e.g., mesh size). Use the following information as a guideline for fish sampling:
  - Use a bongo net to sample egg, larvae, and small juvenile fish; may be use under ice
  - Use small midwater trawls (e.g., 3 m) to sample larvae, juvenile, and adult fish in waters 5-8 m deep from a skiff
  - Use a beach seine (e.g., 37 m) to sample larvae, juvenile, and adult fish in shallow waters from the shore or from a boat. The seine may include a lead-line to allow for contact of the seine with the substrate, and a float-line to allow it to float on the surface
- For beach seine sampling (larvae, juvenile, and adult fish) (4 people recommended) (see photograph)
  - Standard NOAA methods for beach seines are to set the net as a "round haul" by holding one end on the beach, backing around in a skiff with the other end in an arc back to the beach about 18 m from the starting point, and pulling the seine onto shore

  - When sampling by wading:
    - With one person holding each side of the seine, walk perpendicular to the shoreline (starting at the deepest accessible water) pulling the seine through the water. Alternatively, hold one brail stationary at the shore, and pull the other brail in a perpendicular position. Walk the end of the seine towards the shoreline following an arch. Make sure the lead line maintains contact with the bottom during the haul
    - Depending on the length of the seine, the seine can be lifted up prior to reaching the shoreline. Be careful not to drag or to snag the net as fish can be killed or escape
  - Quickly transfer seine contents to clean buckets filled with clean seawater at ambient temperature and, if possible, equipped with an aerator. This is important if samples are not going to be processed in a timely manner
- Fish traps can also be used to collect adult fish. The opening size and mesh size will determine what size fish are captured. Traps should be baited – preferably with a clean piece of bait fish or commercial fish bait pellets. Traps can be left on the bottom, hanging mid-water or deployed under ice and should be checked regularly. Fish traps cannot be used to estimate diversity or abundance, but can be used to capture fish for chemical analysis or other purposes.
- Identify fish to species (or family) and tally the number of each species (or family). Measure the fork length (FL) and/or total length (TL) of up to 50 individuals of each species. If time allows, also weigh these 50 individuals. Record numbers of dead fish by species (or family).
- Prior/during fish sorting:
  - Label the sample tray with the sample ID code in large print and take a photograph of the tray containing the fish sample
  - The sample tray will be sorted into smaller, water-filled trays by major species (or families)
  - Process each sample as quickly as practical to reduce stress to fish that will be released
  - Sort specimens (identified at the lowest possible taxonomic level or by family) from the collection buckets into water-filled trays, only keeping the targeted species (if indicated in the sampling plan)
  - Count, weigh, and measure specimens from each tray, and collect a subsample for further analysis:

    - For chemical analysis: place samples in a glass jar (for small fish or tissue samples) or wrap the fish in two layers of pre-cleaned aluminum foil and place in a zip-loc bag (for larger fish) and store in a cooler (do not freeze)
    - For other analyses: fix samples in jars, plastic vials or buckets containing 10% buffered formalin in seawater, and keep cold. Subsamples may be preserved in ethanol for specific analyses. It is ok to keep several species in the same bucket
- For midwater trawl sampling (larvae, juvenile, and adult fish):
  - Use a small midwater trawl mounted with a 0.5-mm mesh plankton net. Refer to the Plankton guidelines for details
  - Lower the net to the appropriate depth in a vertical position and raise it against the direction of the current at a continuous rate of 0.5 m/s to minimize avoidance of the net by fast-swimming fish
  - At the surface, rinse down the outer sides of the net three times with seawater avoiding the net opening, and do not let the net drop below the water surface
  - After each tow, if the sample contains juvenile or adult fish larger than 0.5 cm follow the fish sorting guideline above. Transfer the contents of the net into a cleaned sample jar
  - Preserve samples with 10% buffered formaldehyde (preferred) or 95% ethanol in seawater
  - Maintain samples at low temperature (6ºC) (preferred), but do not freeze
  - Rinse the net with clean seawater between sites. Avoid sampling near sediments and macrophytes
  - Collect a subsample for chemical analysis as described above
- When sampling fish (fish eggs and newly hatched larvae) under ice sheets:
  - Clear loose ice and snow away from the sampling location and drill through the ice
  - Clean the drill hole area from potential sources of contamination, and allow several minutes for the water to flow freely under the ice before taking a sample
  - Carefully lower the bongo net(s) and follow the rest of the sampling procedure and preservation as described above
  - Fish traps can be deployed below ice to capture adult fish – see guideline above
- To collect fish bile samples for PAH and PAH metabolite analysis:
  - Bile samples must be collected from freshly sacrificed fish, within 20 minutes of their death. Minimum fish size is approximately 5 cm
  - Large fish can be sacrificed by administering a moderate blow to the nape using the fiberglass fish club. Take care to strike the fish with some restraint to avoid bleeding or other fluid losses
  - After sacrificing each animal, open the body cavity using scissors (see photograph). Use one set of tools to cut open the animal and a separate set for cutting tissue inside of the animal
  - Record species, gender, and length for each fish that a bile sample is obtained from (use Tissue Sample Collection Form)
  - Separate the gall bladder (sac-like organ that is green to yellow in color) from the liver. Be sure to grip the gall bladder by the bile duct to prevent bile loss (see photographs)
  - If there is blood on the outside of the gall bladder, rinse it with distilled water
  - Hold the gall bladder at the mouth of the 4 mL amber glass vial and puncture bladder with the scalpel blade
  - Collect as much bile as practical, at least 50 µL. In smaller fish, it may be necessary to composite bile from multiple fish of the same species

  - Use clean dissecting equipment or decontaminate equipment between each fish
  - Keep all bile samples on ice during transport

- Sub-samples of representative species may need to be collected from each tow and stored for analytical chemistry analyses of tissues (PAHs; composite 30-100 g of wet tissue weight). To avoid contamination, collected fish should be kept intact, wrapped in two layers of precleaned aluminum foil, and placed in plastic bags. Collect at least 5 individual samples per target species per sampling station. Follow general guideline provided in the Shellfish Tissue guideline.
- Record the presence of oil, weather conditions, etc. in field notes.

Sample Labeling and Record Keeping

- Verify that all samples are properly labeled, and that field sample forms are properly filled out.
- Follow chain of custody procedures for securing samples and complete chain of custody forms (See Chain of Custody guidelines).
- Complete the Chain of Custody form, noting where each sheen sample was collected, sampling equipment used, time/date of collection, size and container type, and sampler name.
- Make special notation on the Chain of Custody form about any problems or observations during sampling.
- Maintain strict chain of custody during sample storage and transportation.
- Record the sample number on both the sample jar label and lid. Record the following on the field sample form:
  - Sample collection site (NRDA sample grid ID and GPS coordinates)
  - Sample matrix (fish, bile)
  - Sample #, date/time, tidal elevation, water depth
  - Species collected, type (live/dead), number of individuals, size range, sample type (whole, bile)
  - Describe the oiling conditions on the adjacent shoreline (using standard shoreline assessment terminology), weather conditions, sediment characteristics, presence of biota, vegetation or debris, odors and other relevant information on the field data sheet
  - Sediment characteristics: grain size, texture, color, biota, vegetation, debris, odor, etc.; vertical changes in sediment characteristics
  - Characteristics of suspended material in the water sample: turbidity, texture, color, biota, vegetation, debris, odor, etc.
  - Record observations of any external evidence of contamination
- All sample numbers must be unique. Use the sample number convention provided by data management if available. Otherwise, the sample number should consist of a sample team ID and sequential numbers. For example AKA-0001, AKA-0002, etc.

- Documenting oil exposure is best accomplished with photography, video, and good field notes and sketches using standard shoreline assessment methods.
- Keep a detailed photo log so that each photograph can be labeled.
- Note any deviations from the recommended guidelines in the field book.

Sample Preservation, Recommended Holding Times and Shipping

- Follow chain of custody procedures for sample storage and shipping.
- Ship highly oiled samples separate from lightly or unoiled samples to reduce risk of cross-contamination.
- Depending on the remoteness of the sampling location, processing for shipping may not be achievable. If this or any other issues are anticipated, freeze samples that can be frozen as soon as practical and ship samples to the laboratory following special shipping required to maintain samples in a frozen state.
- Immediately following collection, place all fish samples preserved in formalin in a cooler. DO NOT FREEZE.
- Fish samples collected for chemical analysis and bile samples should be place immediately in coolers and kept at 4°C. Freeze as soon as practical. These samples can be stored at -20°C for a year or more.
- Refrigeration temperature shall be recorded upon sample storage and monitored and recorded periodically to ensure proper refrigeration.
- Do NOT use freshwater when sampling and preserving samples collected in salt water.
- In below-freezing temperatures, collapsible jugs of warm water can be used in the cooler between fish samples to prevent them from freezing.
- Preserve fish samples immediately after collection, and discard samples not preserved within three hours of collection.
- Tape lids on sample bottles so that they do not accidentally come off.
- Use packing material, such as bubble wrap or sorbent pads, around glass jars to prevent breakage during transport and shipping.
- Ship samples directly to the laboratory as soon as practical, overnight (preferred), with completed Chain of Custody forms. If necessary, samples can be stored under specified conditions and with complete chain of custody until they can be shipped. Assure that samples are packaged to protect them from breakage, shipping containers are sealed and use ice packs or dry ice to maintain storage temperatures during shipment to the lab.
- NEVER discard any samples even if these have exceeded their recommended holding times or storage temperatures.

Sample Volume

- At least 50 µL of fish bile from a single fish or composite from multiple fish of the same species.

Analytical Methods

- Refer to those under Water and Shellfish Tissue guidelines, if applicable.

Key References

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Appendix A

Unique field data forms may be included in the work plan if one has been developed, otherwise, use the attached form.

- Adapt the form as needed prior to use.
- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Make any additional notes that do not fit on the form in a field notebook and indicate the presence of associated additional notes on the field data form.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Electronic versions of field data forms are available. Coordinate data entry with NRDA data management personnel.

Attached Forms:
- Fish Field Data Sheet
- Tissue/Wrack Sample Collection Form
- Fish Sample Collection Form

Fish Field Data Sheet

1 Date: _____ Time: _____ Site ID: _____ Team ID: _____

2 Data recorder / Affiliation: _____

3 Other team members / Affiliation: _____

4 Site characterization

Site Waypoint # _____ Latitude (DD): _____ Longitude (DD): _____

Sampling method (check one): _____ Beach seine _____ Other: _____

Habitat (circle one): Nearshore / Lagoon / Estuary

Benthic Habitat (circle one): Eelgrass / cobble / sand bottom / tundra / other:_____

Water depth (m): _____

Weather conditions: _____

Oiling conditions: None / Sheen/ Light / Moderate / Heavy

Photos: _____

5

Species	Total Number Counted	Number Collected/Preserved	Notes

6 **Sample collected for chemical analysis: yes / no**

Notes:

Measurements of 50 individual fish

7	Species	Length (mm)	Standard Length (SL) Total Length (TL) or Fork Length (FL)	Weight (g)	Notes

Sample Collection Form - TISSUE/WRACK										
Lead Sampler's Name/Phone								**Sampler Team Code**		
Lead Sampler's Affiliation								**Resource Group**		
NRDA Contact/Phone								**Resource Group Leader**		
Incident Name								**Habitat (e.g., sand beach)**		
General Location Description								**Sample date (mm/dd/yyyy)**		
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Species (NA for Wrack)**	**Tissue Type (NA for Wrack)**	**Number in sample (NA for Wrack)**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**
NRDA Sample Grid ID	*(T)issue or Wrack (R)*	*Sample # and A, B, or C for portion of composite*	*(24-hr clock, local time)*	*Species collected*	*Whole or tissue type*	*Number of organisms in sample*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD - YY'Y.YYYYYY*	*Description of sample, including size (weight, length), equipment, photos numbers, etc.*
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)										
Sample relinquished by:				**Received by:**						
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**			

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball or Water	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H)	(GR)ab (SC)rape (OT)her	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA <for Wrack only>	
Sample Identifier system			
Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

Sample Collection Form - FISH											
Lead Sampler's Name/Phone								Sampler Team Code			
Lead Sampler's Affiliation								Resource Group			
NRDA Contact/Phone								Resource Group Leader			
Incident Name								Habitat (e.g., sand beach)			
General Location Description								Sample date (mm/dd/yyyy)			
Location Code	**Matrix**	**Sample Number (two digits)**	**Sample Time**	**Sampling method**	**Sample position/ Depth**	**Sample Size and Units**	**Sample QA/QC Type**	**Latitude**	**Longitude**	**Sample Notes**	
NRDA Sample Grid ID	*(FI) Fish*	*Sample #*	*(24-hr clock, local time)*	*Method of sampling (seine, bongo etc.)*	*Description of where sample was taken in water column*	*Volume of water sampled*	*Normal sample or Field QA/QC type*	*Latitude in DD XX.XXXXXX*	*Longitude in DD-YY'Y.YYYYYY*	*Description of sample, including equipment, photos numbers, etc.*	
Survey Notes - (weather, wildlife, field team composition, sampling design changes, photos, etc.)											
Samples Relinquished by:				**Received by:**							
Date	**Time**	**Signature - Field Sampler**	**Print Name- Field Sampler**	**Date**	**Time**	**Signature - Sample Runner/ Command Post**	**Print Name - Sample Runner/ Command Post**				

Matrix	Sample methods and descriptions		Sample Area Sketch
Sediment or Soil	**Sampling Method**	**Depth units**	
(S)ediment Soil (L) Blan(K) Water	(GR)ab (CO)re	(c)m (m) (i)nches (f)eet	
Oil, Tarball or Water	**Sampling Method**	**Sample Position/Depth**	
(O)il Tarball (B) (W)ater Blan(K) Water Other (H)	(GR)ab (SC)rape (OT)her (N) Net	(FLOAT)ing (SUB)merged (STRAND)ed (COV)ering 0 - (Surf)ace <depth in meters> m	
Tissue or Wrack/ Plankton or Fish	**Tissue Type**	**Tissue Type (Continued)**	
(T)issue Wrack (R) Blan(K) Water (P)lankton (FI)sh (SH)ellfish	(WH)ole body Whole body w/o shell (WNS) Chorioallantoic Membrane (CAM) Egg (EM)bryo Fillet with skin (FS) Fillet without skin (FWOS) Gall Bladder (GB) Leaves (LEV) Leaves and stems (LVS) (LI)ver	(MU)scle Yolk NA <for Wrack only> **Species** <enter species> NA	
Sample Identifier system			
Sample IDs : Team ID-Sequential Numbers (ex. AKA-0001)			

OTHER SUPPORTING GUIDELINES

Guidelines for Collecting Ephemeral Data in the Arctic: ALTERNATIVE EQUIPMENT/METHODS

September 2014

Note: The media- and resource-specific guidelines detail the preferred equipment and methods for collecting ephemeral data. If the preferred equipment is not available and/or the methods are not feasible, this guideline provides alternatives that may still produce usable and legally defensible data. These guidelines do not necessarily consider all possible scenarios under which samples may be collected. Use best professional judgment to modify these guidelines according to site-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on alternative sampling equipment and methods to be used when the preferred equipment and methods are not available or feasible. The guidelines will help ensure the integrity, utility, and defensibility of samples collected to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations using alternative equipment or methods.

General Guideline

- Follow a sampling plan/work plan if one is available.
- Use the preferred equipment and methods detailed in the media- or resource-specific guidelines to the extent possible; only use alternatives when there are no preferred options.
- If possible, consult with the NRDA coordinator or lead scientist prior to utilizing alternative equipment or methods.
- Availability of sampling equipment should be taken into account when prioritizing ephemeral data collection.
- Maintain normal quality assurance and quality control (QA/QC) measures detailed in the media- and resource-specific guidelines – additional QA/QC may be required to support samples collected with alternative equipment or methods.
- Follow normal chain of custody procedures (see Chain of Custody guideline).
- To the extent possible, use the same sampling volumes, areas, timing, and procedures described in the original media- and resource-specific guidelines.
- Samples collected with alternative equipment or methods should be labeled, stored, and shipped in accordance with the original media- and resource-specific guidelines unless otherwise noted.
- All deviations from standard guidelines should be noted in the field data sheets or field book.
- Photographs or other documentation of alternative equipment or methods are desirable.
- Use of appropriate personal protective equipment is always essential.
- ***Never throw out samples!***

Containers

Alternatives to certified as organic-free (i.e., solvent rinsed) glass jars with Teflon-lined lids for sampling and storing liquids (e.g., water, oil, etc.) for chemical analysis.

Possible Alternatives	Cleaning or Preparation	Quality Control	Notes
Preferred alternative A: Laboratory cleaned glass jars with lids.	Clean jars and lids with dilute detergent (Liquinox or similar), rinse with distilled water followed by deionized water. Rinse jars and lids three times with pesticide-grade or better hexane and acetone. Bake the glassware in a laboratory oven (if available) or let the lids and glassware dry in a clean hood. If lids are not Teflon-lined, put a piece of clean aluminum foil over the top of the jar before screwing on the lid.	Cleaning should be done by a chemist, in a laboratory, following a Standard Operating Procedure (SOP), and with appropriate documentation. Source of glassware should be noted.	If new, certified organic-free clean glass jars are not available, used jars that have been organic cleaned are the best alternative. If using aluminum foil to isolate the sample from the plastic lid, assure that the jar is completely sealed and not leaking.
Preferred alternative B: Laboratory glass jars of any shape or volume with lids.	Clean jars and lids by washing and solvent rinsing as described above. If hexane and/or acetone are not available, wash the jars and lids with dilute detergent and rinse with distilled water. If lids are not Teflon-lined, place clean aluminum foil over the top of the jar before screwing on the lid.	Send at least three clean, empty, sealed jars along with the field samples to the lab for analysis of possible contamination.	Organic solvents should be used only under a chemical hood or in a well-ventilated area. If using aluminum foil to isolate the sample from the plastic lid, assure that the jar is completely sealed and not leaking.
Other alternatives: Glass jars (e.g., canning jars) with plastic or metal lids.	Unused canning jars are preferable. Clean jars and lids as described above. Use aluminum foil under the lid to isolate the sample.	Send at least three clean, empty, sealed jars (of each type used) along with the field samples to the lab for analysis of possible contamination.	Only jars with lids that seal tightly should be used (e.g., canning jars with lids with gaskets). If jars leak, there is a risk of cross contamination. Assure that the jars are sealed and not leaking before transport, storage, and shipping. Use durable jars that will withstand shipping in coolers.

General notes:

- DO NOT use plastic bottles of any kind to store samples collected for chemical analysis.
- Always package glass jars for transport, storage, and shipping in a way that will prevent leaking or breaking.
- It is preferable to meet or exceed the minimum sample volume requirements noted in the media-specific guidelines.
- To the extent possible, use jars that approximate the sample volumes needed.
- Follow media-specific guidelines about head space, air content, storage conditions, and recommended holding time.

Alternatives to certified as organic-free (i.e., solvent rinsed) glass jars with Teflon-lined lids for sampling and storing solids (e.g., sediments, tar balls) for chemical analysis.

Possible Alternatives	Cleaning or Preparation	Quality Control	Notes
Preferred alternative A: Laboratory cleaned glass jars with lids.	See above (alternatives for liquid samples).	See above.	See above.
Preferred alternative B: Laboratory glass jars of any shape or volume with lids.	See above (alternatives for liquid samples).	See above.	See above.
Other alternative: Wrap sample tightly in clean aluminum foil and place in a zip-lock bag.	Aluminum foil must be unused. Rinse foil with hexane and acetone if available. Place sample in aluminum foil (shinier side facing out), wrap completely and fold foil to seal. Place the sample in a clean, unused zip-lock bag or other clean bag depending on the size of the sample.	Samples will need to be packed in a tamper-proof container, such as a box or cooler, for chain of custody procedures.	This method is only appropriate for solid samples that will not leak out of the aluminum foil. Solid samples wrapped in aluminum should be sealed in an air tight bag or container to prevent vaporization or cross contamination. Package samples for storage and transport to avoid puncturing or tearing the aluminum foil or bag.
Other alternative: Glass jars (e.g., canning jars) with plastic or metal lids.	See above (alternatives for liquid samples).	See above (alternatives for liquid samples).	See above (alternatives for liquid samples).

General notes:

- DO NOT use plastic bottles of any kind to store samples collected for chemical analysis.
- When using plastic bags, assure that the sample is completely isolated for the plastic.
- Always package glass jars for transport, storage and shipping in a way that will prevent breakage.
- It is preferable to meet or exceed the minimum sample volume requirements noted in the media-specific guidelines.
- Consider if frozen samples will remain solid if subject to temperature changes during transport, storage, or shipping when selecting an appropriate container.
- Follow media-specific guidelines about storage conditions and recommended holding time.

Cleaning

Alternatives to using organic solvents for cleaning glassware and sampling tools.

Possible Alternatives	Cleaning or Preparation	Quality Control	Notes
Preferred alternative: Detergent and deionized or distilled water.	Clean with dilute detergent, rinse with clean water followed by a final rinse with ultrapure or deionized water (ideal) or distilled water.	Take rinsate blanks from equipment that is being washed and reused during sampling and more frequently when working at oiled sites to test for any possible cross contamination. Note on the field data sheets when and where rinsate blanks were taken.	Whenever possible, clean glassware and other equipment before using it in the field. It is preferable to use disposable sampling equipment, if available.

General notes:

- Bring clean glassware and disposable sampling equipment into the field to minimize the need for cleaning in the field.
- For equipment that must be reused, obtain samples from unoiled locations before oiled locations as much as practical to minimize risks of cross contamination.

Storage Conditions

Alternatives to temperature-controlled sample storage when a refrigeration or freezer (-20°C) unit does not have an auditable temperature record.

Possible Alternatives	Cleaning or Preparation	Quality Control	Notes
Preferred alternative A: Programmable temperature logger (TidbiT or similar). Use to verify that storage temperatures are within the range specified for the samples.	Program the temperature logger to take readings at regular intervals (e.g., every hour). Place logger with samples in cooler, refrigerator, freezer, or other storage unit.	Record logger serial number and other relevant information. Note when and where loggers were located and document any occurrences that may have caused anomalous readings. Data from the loggers should be downloaded and copied to an uneditable format.	Assure that temperature logging intervals do not exceed the memory or battery life. Temperature loggers may be maintained in the storage unit or with the samples depending on storage requirements and shipping methods.
Preferred alternative B: Storage stability samples. Use to verify the stability of samples stored at temperatures outside of the specified range.	Obtain or prepare blank matrix spikes. Alternately, a known quantity of a mixture of labeled analytes can be overspiked on samples collected in the field. Chemical preparation must be done by a chemist with knowledge of the work plan.	This is a quality control measure – obtain at least three replicate storage stability samples.	This should be done by or in very close consultation with a chemist. Spiked storage stability samples or chemicals for overspikes must be prepared by a chemist in a lab. This should be used if it is not possible to follow the sample storage condition guidelines during storage or shipping. A temperature logger should also be used (see above).
Other alternative: Manually record the temperature.	Place a thermometer with the samples in a cooler, refrigerator, freezer or other storage unit and record the temperature at regular intervals (e.g., every hour). High/low thermometers can also be used.	Maintain accurate records of when the readings were taken and who took them.	Never dispose of samples, even if a recorded storage temperature falls outside of the specified range.

General notes:

- If temperature-controlled refrigeration or freezer units are not available, it is important to store the samples at a temperature as close to the one specified in the guidelines as practical.
- This can be achieved by maintaining the samples in a cooler with ice packs or jugs of warm water (depending on the desired storage conditions and ambient temperatures).
- Under the appropriate conditions, samples can also be buried in the snow or left in a cooler subject to ambient temperature.
- Consider how to maintain chain of custody if samples are held in an unsecured location.

Holding Times

Alternatives for documenting storage stability if recommended holding times exceed those specified in the methods.

Possible Alternatives	Cleaning or Preparation	Quality Control	Notes
Preferred alternative: Storage stability samples *Use to verify the stability of samples stored for longer periods of time than specified before analysis.	Obtain or prepare blank matrix spikes. Alternately, a known quantity of a mixture of labeled analytes can be overspiked on samples gathered in the field. Chemical preparation must be done by a chemist with knowledge of the work plan.	This is a quality control measure – obtain at least three replicate storage stability samples.	This should be done by or in very close consultation with a chemist. Spiked storage stability samples or chemicals for overspikes must be prepared by a chemist in a lab. This should be used if there is no possibility of following the sample storage condition guidelines during storage or shipping. A temperature logger should also be used (see above).
Other alternative: Post sampling storage stability study.	In order to do a storage stability study, the conditions that the samples were kept in must be well documented. Use a temperature logger or other method to monitor storage conditions. Document other relevant storage conditions.	Appropriate QC should be maintained during the storage stability study.	Note any apparent changes in the original sample that may be indicative of storage stability issues, for example, volume loss or color change. A post-sampling storage stability study would be conducted in a laboratory by a chemist.

Other

Alternatives for other materials needed to ensure the quality and legally defensibility of samples collected in the field (not an extensive list).

Material	Preferred Alternative 1	Preferred Alternative 2
Evidence tape	Duct or gorilla tape	Vinyl electrical tape
Amber glass jars (to protect samples collected for chemical analyses from sunlight).	Aluminum foil wrapped around the glass jar.	Newspaper wrapped around the glass jar. Maintain samples in cooler, box or other storage container protected from light exposure.
Formalin (for sample preservation)	Ethanol.	Methanol.
Waterproof paper (for printing field sampling forms).	Regular paper, plus zip-lock bags to keep all forms protected from water and moisture.	
PVC quadrats (for quantitative analyses).	String quadrats.	
Collapsible jugs (for storage temperature regulation).	Hand-warmer heat packs.	
Bubble wrap (for packaging samples in glass jars).	Sorbent pads.	Newspaper.
General notes: • Note any deviation from the preferred alternative in the field note book.		

Guidelines for Collecting Ephemeral Data in the Arctic: CHAIN OF CUSTODY

September 2014

Note: Chain of custody procedures should be followed to ensure the integrity of all samples collected to support a Natural Resource Damage Assessment (NRDA) case.

Guideline Objectives

The primary objective of this document is to provide guidelines on chain of custody procedures for ephemeral data and samples collected in the field during the early stages of an oil spill in the Arctic to support NRDA exposure and injury evaluations.

Background

Chain of custody procedures are followed to authenticate a sample from the time it is taken until the results are introduced as evidence. For the purposes of litigation, agencies must be able to prove the legal integrity of all samples and data introduced as evidence. This means that it is necessary to have an accurate written record to track possession, handling, and location of samples and data from collection through reporting. Chain of custody facilitates this verification process. Failure to follow chain of custody procedures in this guideline does not necessarily render data unusable; however, any deviations from the chain of custody guidelines should be noted. Assuring that proper chain of custody guidelines is followed is vital to assuring the integrity of the samples, and the data generated by the analysis of those samples.

Responsibilities

All samplers handling samples collected for NRDA MUST follow this procedure when collecting, handling and securing samples. All team leads and supervisors are responsible for ensuring that the designated custodian(s) understand this procedure and strictly adhere to it for all sampling events.

Important Definition

- **Chain of Custody Form**: A document detailing who is legally responsible for samples at any point in time from collection until the sample is received by the laboratory.
- **Custody**: A sample is in your custody when
  - It is in your actual physical control and presence
  - It is in your view after being in your possession
  - It is not in your physical presence, but is secure in a place of storage to which only you have access
  - It is not in your view or physical presence, but is secured in a place of storage or secure area to which only you and identified others have access
- **Secure Area**: An area in which entry is restricted by keyed lock or similar to a designated custodian

Equipment for Chain of Custody

- Sample labels
- Tamper resistant evidence tape (for small sample jars and large shipping containers)
- Permanent markers
- Chain of custody and field data forms
- Secure storage area

Sample Collection – Chain of Custody

Note: As few people as practical should handle the sample from when it is collected through laboratory analysis.

- Sample custody begins immediately after a sample is collected. The sampler who collected the sample is responsible for the preservation and integrity of the sample(s) until that responsibility is transferred to someone else, and documented with the chain of custody form. This chain of custody form then travels with the sample(s) and is used to document any other transfers of custody.
- When a sample is taken, the sampler must:
  - Complete a sample tag or label that identifies each sample. Use waterproof ink and attach the label to the sample jar or container at the time the sample is collected. Labels should contain that following legible information:
    - Sample number
    - Sample type (e.g., sediment, water)
    - Sample containing hazardous goods (such as formalin used as a preservative) (if applicable)
    - Time/date of collection
    - Location
    - Sampler name(s)
  - Seal each sample jar or container with tamper resistant evidence tape. When sealing jars, the tape should connect the jar to the lid. The sample collector should sign and date evidence tape so that the signature is partially on the tape of both the lid and the jar
  - If the sample is collected in a container that is not tamper proof (such as a plastic bag) then the container should be sealed with tamper resistant tape, a serial numbered zip-tie, or other means of verifying that the container has not been opened. The sample should also be stored in a container that is appropriate for chain of custody, such as a box or cooler that can be sealed with tamper resistant tape and signed by the collector
  - If tamper resistant evidence tape is not available, use masking or duct tape and sign across the end of the tape
  - Enter each sample on the chain of custody form
  - Document the sample in the field data sheet, noting details about the sample that may be pertinent later during sample analysis and injury determination
- The sample collector is responsible for care and custody of the samples until they are turned over to an assigned custodian or properly dispatched to the receiving laboratory. All custodians must assure that each sample remains in his/her custody (as defined above) so that no one can tamper with it during the entire duration of their responsibility.
- When samples are turned over to a new custodian:
  - The current custodian must officially relinquish the samples by signing the chain of custody form
  - The new custodian must review the samples, ensuring that they are in good condition and that the sample IDs matches the chain of custody form. Any damage or deviation must be noted on the chain of custody form before the new custodian accepts the samples by signing the form
  - The former custodian must retain a copy of the full set of forms. The original chain of custody forms always stay with the samples
- Pack and seal samples in suitable containers to avoid damage. A sample seal should be attached across the lid of each shipping container in such a manner that the container cannot be opened without breaking the seal. This lock and/or seal are not to be removed until the shipping container is opened by the laboratory custodian or designee.
- Ship samples by registered courier. Other certified shipping services may also be used. Keep all shipping receipts as part of the permanent chain of custody documentation regardless of how samples are shipped. The shipper does not need to sign the chain of custody form.

- Couriers picking up samples at the airport, post office etc. should sign the shipping documents to acknowledge receipt of the samples.

Photographs

- Digital photographs can be used as evidence. Like with physical samples, the objective is to be able to ensure that the photographs are an accurate representation of what was seen in the field (see Field Photography guideline).
- It is important to protect the legal integrity of digital photographs stored on digital memory cards (SD cards), as well as the legal integrity of the SD card itself.
- Digital photographs taken in the field and information stored on a digital memory card or camera internal memory MUST NOT be deleted, no matter the quality or other issues that may arise.
- All digital photographs files MUST be stored sequentially on the SD card and not renamed.
- Photographs should be downloaded to a secured PC and copied to an un-editable format (e.g., CD/DVD).
- Always keep a back-up copy of all photographs.
- See the Field Photography guideline for more details.

Other Important Considerations

- **Custodian list**: Maintain a list of people who are custodians on samples that includes how each person is related to the assessment.
- **Multiple custodians**: If multiple qualified custodians have access to a secure sample storage area then it is not necessary to document change of custody between custodians. All custodians should be identified on the custodian list.
- **Chain on multiple sheets**: Starting chain of custody documentation on one form and continuing it on a second form for the same samples is not a break in the chain. Care must be taken to keep the forms together.
- **Broken chain**: If the chain is broken for any reason or if you foresee deviations from the procedures in this guideline, contact the legal team for guideline. Changes to the procedures can be made but should be informed by a lawyer. Do not discard ANY samples even if the chain of custody has been broken.
- **Samples on the same sheet are split**: If samples that were recorded on a single chain of custody form are split for shipping, clearly mark the original chain of custody form to show which samples were removed, and when and where they are going. Create a new chain of custody form for the samples that are shipped and include a copy of the original chain of custody form. Keep the original form and a copy of the new form with the samples that are not shipped.

Appendix A
Supporting Documentation - Chain of Custody Forms

Chain of custody forms may be provided by the lab that will receive the sample or the NRDA lead, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil, or biro (erasable) ink.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.
- Original chain of custody forms should always stay with the samples. Make a copy of the chain of custody form before sending it with the samples.

Attached form:
- Chain of Custody Form

Chain of Custody Form

Sampler Information		Contact Information
Contact/Phone/Email:		Contact/Phone/Email:
Affiliation:		Sample Questions:
Incident Name:		

Special Instructions:	Analyses requested						# of containers	Lab Name:	
								Waybill Number:	
								Lab Use Only Lab Report #:	
								# of Coolers:	
Turn Around Time:								Cooler Temp:	

Sample ID	Sample Date	Sample Time	Matrix								Comments
	mm/dd/yyyy	*(24-hr local)*		*Enter Analyses above, with preservative specified, if needed. Enter x's in boxes below.*							

Samples Relinquished by				Received by			
Date	**Time**	**Signature**	**Printed Name/Org.**	**Date**	**Time**	**Signature**	**Printed Name/Org.**

Organics Analyses

Aliphatics
Alkylated PAH Homologues
Chlorinated Herbicides (8151)
Dioxins and Furans (8290)
OC Pesticides (8081)
OP Pesticides (8141)
PAH (8270)
PCB Aroclors (8082)
PCB Congeners (680)
Phenols (8041)
PIANO (Volatile Paraffins, IsoParaffins, Aromatics, Naphthenes, & Olefins)
Semivolatile Organics (8270)
Steranes/Triterpanes
Volatile Organics (8260)
[any other analyses or method as needed]
Nutrients (EPA 300.0, 350.1, 353.2, 365.3) - CWVP

Inorganic Analyses

Ammonia
Grain Size
Total Dissolved Solids
Total Kjeldahl Nitrogen (TKN)
Total Organic Carbon (TOC)
Total Solids
Total Suspended Solids
[any other analyses or method as needed]
pH / Salinity (EPA 9040, SM 2520B 18th edition) - CWVP
Soil Organic Matter - CWVP

Petroleum Hydrocarbons

BTEX
Extractable Petroleum Hydrocarbons (EPH)
Petroleum Hydrocarbons (8015)
Saturated Hydrocarbons
TPH Oil & Grease (418.1)
TPH-Diesel Range
TPH-Gas Range
NWTPH-Dx (NW method)
NWTPH-Gx (NW method)
[any other analyses or method as needed]

Metals Analyses

CLP Metals
Low Level Arsenic
Low Level Mercury (1631)
Mercury (7470/7471)
MTCA Metals (As, Cd, Cr, Pb, Hg)
PP Metals (Ag, As, Be, Cd, Cr, Cu, Hg, Ni, Pb, Sb, Se, Tl, Zn)
RCRA Metals (As, Ba, Cd, Cr, Pb, Se, Ag, Hg)
TCLP Metals [any individual metal or list of metals as needed]
Metals (EPA 200.7/6010: Al, Ba, B, Cd, Ca, Cr, Cu, Fe, Pb, Mg, Mn, Mo, Ni, K, Na, Zn) - CWVP

Bio Analyses

Amphipod survival test
Bivalve bioaccumulation test (chronic)
Gonad Condition Index
Infaunal Analysis
Larval Bivalve development test
Larval Echinoderm development test
Length Frequency
Mysid survival test
PCR/DNA
[any other test as needed]
Belowground Vegetation Biomass - CWVP
Aboveground Vegetation Biomass - CWVP
Vegetation Stem Count
Vegetation Longest/Shortest - Stem Length
Vegetation Species ID
Vegetation Live/Dead Sort

Guidelines for Collecting Ephemeral Data in the Arctic: FIELD PHOTOGRAPHY

September 2014

Note: These guidelines are limited data collection aides that do not necessarily consider all possible scenarios under which photographs are taken. Use best professional judgment to modify these guidelines according to area-specific field conditions.

Guideline Objectives

The primary objective of this document is to provide guidelines on procedures for taking photographs and recording videos for ephemeral data and samples collected in the field during the early stages of an oil spill in the Arctic to support Natural Resource Damage Assessment (NRDA) exposure and injury evaluations.

Background

Photographs and video are taken in the field to document the pre-oiling and oiling conditions, and are key pieces of information that can be introduced as evidence. Each photograph should tell a specific part of the story. Before taking a photograph you should consider what critical information you are trying to convey. Did the photograph capture the details you need? Are there key images (data) that you have missed? In the first case, you should take a better photograph. In the second case, you should look for photographs that will fill in the gaps.

Document the Incident, the Location and the Staff

- Photographs are taken to visually communicate what happened at a specific location or sampling site.
- Because photographs can be later viewed by various audiences (e.g., upper level management, Congressional hearings, court, the USCG National Pollution Fund Center, public hearings, training talks, outreach events, etc.) try to capture photographs for all types of audiences.
- Take as many photographs as needed. You may not get a second chance.
- The following is a partial list of subjects to always document:
  - How the oil spill happened – including oil source
  - Oil on the water surface, stranded on the shoreline, and in direct contact or over sensitive areas such as seagrass, shellfish, or kelp
  - Oiled wildlife
  - Oil recovery and cleanup operations (Response)
  - Staff working, sampling (NRDA)
  - Species presence and habitat use
  - Site use by humans (fishing, hunting)
  - Site documentation, such as overviews alongshore and locations of stakes or other site markers
- Time series of photographs are helpful to document exposure or changes in oiling over time. Repeat photographs for a time series by standing in the same spot (using GPS coordinates or a print of the previous photograph).

Document the Injury and Cause of the Injury

- Photographs are an effective way to document injuries caused by oil or response actions, but opportunities to capture these may be short lived. The NRDA team must be prepared to act quickly and decisively.
- Haphazard photography will fail to capture critical information.

Directly Observable Injury

- Photographing and video recording direct injury can be very effective. Take photographs and make videos that clearly show conditions that are or may be caused by oil exposure and response actions, including but not limited to:
  - Oil on biota
  - Dead animals and plants
  - Aberrant behavior (best capture with a video recording)
  - Impacts of cleanup operations
  - Ephemeral evidence of injury: necrosis, bleaching, gaping bivalves, etc.

Causation of Injury

- Photographs and video are good for documenting visible oil exposure and impacts to recreation and human use:
  - When taking photographs of oiled shoreline, include perspective shots that show the degree of oiling as oiling occurs. Repeat day-to-day and tide-to-tide, if possible. Do not rely solely on SCAT to record the presence of oil
  - It is important to document response actions that impact biota (e.g., removing, crushing, re-oiling, hazing) and other resources (e.g., sediment disturbances, etc.)
  - Also document closures of beaches, waterways, access points for fishing or recreation, including but not limited to photographs of official closures (e.g., posted closure signs), congestion effects (e.g., response taking over boat ramps), and popular use areas showing little or no recreational or subsistence use

Qualitative and Quantitative Approach

- Using a systematic photographic process to document oiled areas, reference areas, and the transitions between them can be an effective approach for documenting direct exposure.
- Rigorous photo transect and photo quadrat techniques may be appropriate depending on the assessment.
  - Use the sampling designs used for manual transect and quadrat surveys
  - Include oiled and non-oiled sites, or gradients in a continuum from most heavily oiled to non-oiled

Before Going to the Field

- Use field data forms included in the work plan, if one is available. Otherwise, use forms in Appendix A. Coordinate data form development/modification with the data management group.
- Make sure you have assembled a full photography kit appropriate for NRDA field work (See Appendix A- Full Gear and Field Gear checklists).
- Make sure all photographic gear is ready and complete before going to the field. This includes having:
  - Fully charged batteries
  - Memory cards (SD cards) (see below)
  - Clean lenses

- It is extremely important than the designated photographer and all personnel taking a camera into the field are knowledgeable of key camera settings. The recommended camera settings are as follows:
  - Resolution – MAX
  - ISO – Auto (avoid higher than 400 unless you are an experienced photographer)
  - Mode – Program (P)
  - Time stamp – off, especially if you are using GSP-Photo Link. Unlike film, there is no need to clutter photographs and use up pixel space with a time stamp. That information is automatically recorded in "EXIF" data—which is part of the image file
  - Time – local time
  - Continuous picture numbering – Set to use a running count for file names even after changing or formatting memory cards
  - Daily folders – Set camera to create a new folder each day
  - Advanced settings (e.g., spot metering, custom white balance, etc.) – reset. It is a good idea to return these advance settings to auto or a general setting before you go into the field
  - Camera reset – Most cameras have a way to return all settings to the factory default values. This is useful if images are poor, you have been experimenting with different camera settings and you cannot determine what setting may be causing the problem
- Make sure there are sufficient memory cards. Prior to going into the field, keep in mind the following recommendations:
  - Use high-quality memory cards with large storage capacity. Get enough capacity for a whole day's shooting
  - Be sure all memory cards are working properly and are compatible with the camera being used
  - Changing cards in the field risks getting moisture, salt, and dirt on the memory card contacts and inside the camera
  - Format memory card regularly. It's better to format than to delete all photographs
  - Older cameras may have issues with new cards, but updating firmware may fix the problem
  - At all times protect the electric contacts of memory cards from dirt and mechanical deformation
  - NEVER take out a memory card when the camera is still writing to it. Turn off the camera before changing memory cards
  - Use a quality memory card reader
- Follow this simple recommendations when taking photographs/videos in the Arctic:
  - Battery efficiency is greatly reduced in cold temperatures. Make sure sufficient batteries are taken to the field and, if possible, carry a set of spare batteries close to your body to maintain battery efficiency. When not in use, turn the camera off to save battery life
  - Electronic components of the camera are affected by cold ambient temperatures, but functions return to normal as the camera warms up
  - When snow is present, the camera's exposure meter can be easily confused creating under-exposed images. Set the exposure +0.5 to +1, which will force the camera to expose longer. Use the "snow and ice" mode if available on the camera
  - Never breathe or blow on a cold lens. Instead, dust or brush off snow and debris with a soft cloth
  - NEVER change lenses outdoors when in cold weather or when snow and sleet are present. Moisture or condensation inside the camera can quickly reduce the quality of the photographs and compromise the long-term integrity of the camera
  - While not in use, and prior to bringing cameras indoors after taking photographs in the field, cameras should be placed inside a polypropylene freezer bag, loosely twisted and placed inside the camera bag. Proper storage of the camera will help minimize the condensation that occurs during temperature changes

Learn basic camera functions

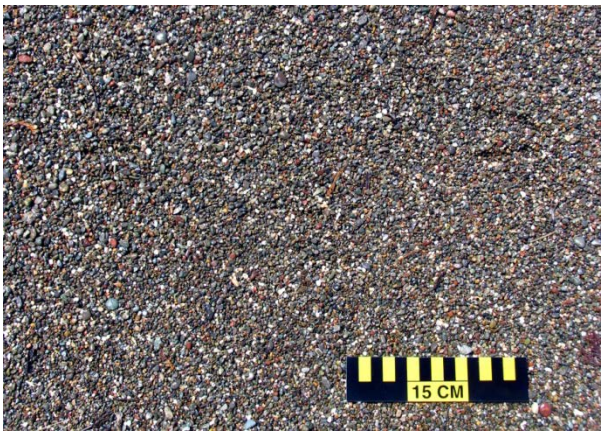
- Remember that each digital camera is different. It is absolutely critical that all camera-users know how to use their assigned camera before going into the field.
- For most field purposes and weather conditions, compact (aka "point-and-shoot") cameras are cheaper, easier to use, more portable, and more resistant to salt, moisture, sand, and other factors. If you are an experienced photographer, can wait for favorable weather conditions, or require photographs for quantitative analyses, single lens reflex (SLR) cameras can provide higher resolution and better quality photographs. SLR cameras generally perform well under freezing temperatures.
- Under some circumstances, and when taking photographs or video of underwater habitats and resources, small drop cameras (e.g., GoPro) or other underwater cameras may be necessary to document these areas.
- Cameras with 7-10 megapixels are recommended.
- The following are some basic functions that everyone should know. Many cameras require you to be in "P" (program) mode (not "A" [auto]) to use these:
  - **Light metering**: Spot. At this setting the camera meters the exposure at a designated spot in the photo frame. Most cameras show the "spot" as a box or circle in the center of the viewfinder. Spot metering is helpful when photographing a subject is much darker or lighter than the rest of the frame
  - **Light metering**: Exposure compensation (+/-) adjustment. This feature tells the camera to make the photograph lighter or darker. It works like the lighter-darker adjustment on most copy machines
  - **White balance adjustments**: White balance settings help the camera adjust the colors in the photographs based on the type of light (fluorescent, incandescent, sunny, cloudy, etc.). Most of the time Auto White Balance (AWB) works fine, but sometimes the camera does not adjust correctly. Manually choosing the type of light can fix the problem
  - **Review photographs**: Know how to use the camera display to review a photograph. Know how to zoom in on the photograph in the display screen to check focus, exposure, and other key details
  - **Forced flash**: In dim light or harsh shadows you may need to force the camera to use the flash to avoid losing details
  - **Continuous shooting**: Most cameras will shoot consecutive photographs while you hold down the shutter. This is sometimes helpful when trying to capture moving wildlife
- Some cameras may have GPS capabilities. The use of these cameras reduce location errors when labeling photographs as the location information is attached to the photo data. Basic GPS capability is essential for all field work, including photography. There are a number of key functions you need to set including (see Appendix A for detail):
  - Local time zone
  - Datum
  - Track (wrap, interval)
  - WAAS (on), etc.

  Note: When the GPS recording is enabled, the camera battery life is shortened considerably.

While in the Field

- At each sampling location or site where photographs are taken, use the GPS to record waypoints. This will help with GPS-photo synchronization and processing (see below).
- At each sampling location or site where photographs are taken, use the GPS to record waypoints. This will help with GPS-photo synchronization and process (see below).

- It is important to take photographs of a sampling site using labeled photo scales (e.g., 15 cm, 6 inches). The photo scale should be in one of the corners, preferably the lower right (see photograph). When necessary because of oiling conditions, disposable scales of standard length, such as wooden tongue depressors, can be used (with proper disposal).



- Scales (and quadrat frames) should have intermediate reflectance, not bright white. A bright scale object can cause the camera to underexpose the rest of the photograph.
- Use spot metering or camera flash to eliminate harsh shadows that can obscure details. Use one of each if you're not sure which is better. Remember that setting the exposure for shadows may wash out and lose detail in bright areas of the photograph.
- Every close up should be followed by one or more wider-angle shots that will show the close up in the context of the rest of the environment. The closer the initial shot the more perspective shots are needed.
- Use the following distances as a guideline:
  - Macro (field of view ≤12"), useful for species identification, fine detail, or injury documentation
  - Close-Up (<1 m^2), useful for general documentation of oiled biota and resources
  - Mid-Level (1-2 m^2 – angled), useful when documenting groups of biota and oiling
  - Distant/landscape (>10 m^2), useful when documenting habitats and spatial patterns of oiling; it is best to have a person in the photograph for scale (see photograph)
- It is important to constantly take photographs in the same sequence to document pre-oiling and oiling conditions, and to keep photographs organized. For example:
  - Start each new location with panorama shots or a narrative video
  - Always photograph subjects from the most close-up to the most zoomed out
- Change batteries before they lose power just as you are taking a critical photograph.
- Use the review feature to ensure that photographs show what you need.
- Use the zoom in function to see if you captured necessary details.
- Note key photographs and important details in the field notebook.
- Record basic information - locations, times, photographer, team members, including descriptions of GPS locations or waypoints.

Taking photographs of quadrats

- It may be necessary to take high-resolution photographs of sampling quadrats for quantitative analysis.
- Quadrats should not be bright white. Make quadrats out of grey PVC or wrap white quadrats in colored duct tape (see photograph).
- All photographs of quadrats must include a label containing the location name, transect, quadrat.

- Take high-resolution vertical photographs of each quadrat, if possible using a tripod or quadrapod, and record GPS coordinates. When taking photographs:
  - Ideally, photographs should be taken during the lowest tide and best light conditions (e.g., closest to midday or when overcast). Avoid shooting into the sun and avoid including sky, ocean, or tidepools in the view
  - High-resolution photographs must include all four sides of the quadrat as these will be used to digitally count individuals and measure their coverage on a computer screen
  - When photographing highly dense quadrats, quadrat frames can be split into 2-sided frames to facilitate computer-based analyses

  - Photographs need to be relatively flat so that the entire quadrat falls within a similar focal plane, with minimal shadowing from crevices or projections. Photographs should be directly perpendicular to the quadrat
  - If possible, use a quadrapod apparatus to support the camera at a constant height (1 m with a 35 mm lens) from the quadrat, and positioned to capture all four corners of the quadrat:
    - A quadrapod consists of a gray PVC or gray Schedule 80 PVC pipe frame with a photoplot-size bottom (0.5 m^2 or 1.0 m^2 internal dimensions) connected by 4 poles to the frame supporting the digital camera
    - Strobes mounted laterally and away from the camera can enhance lighting of the quadrats and reduce shadows
  - The best quality photographs are obtained by optimizing the ISO, aperture, and shutter speed
  - Remember that all quadrat images must be of sufficient quality to allow a positive identification and enumeration of the species in the quadrats

Taking panoramic photographs

- Panoramas are often un-necessary but if you need a wide, detailed photograph do the following
  - Keep photo edges parallel
  - Do not change “zoom” factor
  - Overlap photographs by about 30%
  - Place a scale or natural distinctive feature in each overlap area for accurate alignment
  - Do not move the photo scale
  - Use manual mode to set shutter and aperture if you are comfortable with this
  - Note which photographs are part of the panorama
  - Lock your elbows against your sides for stability and pan as close to horizontally as you can. Use a tripod or monopod if you have one

Taking video

- A short video synopsis of a location can be very helpful later for relaying or reviewing the general layout of a location.
- It is important to take video recordings documenting ice scour, presence of ice, wave regimes, etc. as these can have impacts on oil fate.
- Take 30-45 seconds to slowly pan through a site while narrating key features.

Taking photographs while flying

- Taking photographs from a plane or helicopter can be difficult and requires additional skills. Point and shoot cameras can take good photographs from the air but SLR's typically perform better.
- When taking photographs from a plane or helicopter:
  - Do not wear bright clothing as these may reflect in the windows of the aircraft
  - Use manual focus to set cameras to infinity (∞). This avoids accidentally focusing on the window
  - Using image-stabilized cameras or lenses will help take good quality photographs
  - To prevent transmitting aircraft vibration to the camera, do not rest the camera on an aircraft window frame or other part of the aircraft structure. Instead, hold the camera with your arms braced against your legs or torso, or the camera held against your face
  - Avoid shooting through a bubble window
  - Smaller aircraft often have sliding windows, or easily removable windows or doors (see photograph). Make arrangements with your pilot before take-off
  - Avoid taking photographs towards the sun
  - Consider using one zoom level. Survey flights often are directed to maintain a specific altitude. By maintaining a constant zoom level you will be able to compare items in successive photographs. Remember there are no scales in aerial photographs
  - Record on the field notebook the basic flight plan including altitude and distance from shore, aircraft type, pilot and passenger names, port or starboard

Taking underwater photographs

- When taking underwater photographs:
  - Ensure that the camera is set on the underwater mode, which is design to filter some wavelengths
  - If possible, include a scale with each photograph (described above)
- Underwater photographs or video may be the only form of documenting underwater quadrats. Take photographs/video of each entire quadrat from an angle as vertical as practical. Photographs or video should include:
  - The general station location and setting, showing permanent stakes (if any)
  - Examples of subtidal resources and habitats (e.g., kelp field, eelgrass bed)
  - Sites where samples were collected
  - Representative examples of the extent and degree of oiling
  - Examples of services provided by subtidal resources and habitats
- Underwater cameras may be the only way to document impacts, if direct sampling of subtidal resources is unpractical or unsafe. A high-definition underwater camera can be deployed from a vessel, but this type of sampling requires previous training. Briefly:
  - Mount the underwater camera in a 'down-looking' orientation on a towfish deployed directly off the stern of the vessel

  - Allow the camera to follow the contour of the desired subtidal habitat
  - Maintain the field of view as constant as possible (1 m^2)
  - The vessel speed should be held as constant as possible (about 1 m/s) to facilitate estimation of distances
  - Conduct straight-line underwater video transects (randomly selected) perpendicular to the shoreline and encompassing the width of the subtidal habitat (e.g., kelp field, eelgrass bed)
  - Carefully catalogue all underwater videos and ship to the appropriate laboratory for processing and interpretation
- Underwater cameras may be the only way to document impacts to water column resources underneath ice sheets. A high-definition underwater camera can be deployed from vessel, but this type of sampling requires previous training. Briefly:
  - Mount a small drop camera (GoPro or other underwater camera) on a pole and send down a hole on the ice to take pictures of the surrounding ice and ice/water interface
  - Carefully catalogue all underwater videos and ship to the appropriate laboratory for processing and interpretation

Upon Returning from the Field

Legally defensible photographs

- Creating a legally defensible photograph record requires:
  - Maintaining a complete photograph record. DO NOT delete photographs from the camera or from your computer before the official archive is created
  - Keep one set of photographs that are never opened. In practice this means transferring one copy of the photographs from the camera memory card to a computer and then to a DVD-R or CD-R (non-editable) without ever opening them. The resulting continuous set of photograph files that have not been opened will demonstrate that that you have a full, un-edited, photograph record for the court
- When return from the field download all photographs to a computer. Before reviewing photographs on the computer (review = open):
  - Create a copy in the "*Working*" directory and one copy in the "*Archive*" directory
  - The "*Working*" directory is used to process photographs through GPS-linking software and to log all photographs. DO NOT rename files in the "*Working*" directory
  - The "*Archive*" directory MUST include unopened, un-editable copy of all photographs. Burn the "*Archive*" directory to DVD-R. Do this when you have enough to fill a CD/DVD or at some set interval (every 2 days), and make a copy of the CD/DVD
  - NEVER open the files stored in the "*Archive*" directory
  - Make additional backup copies can be made to portable hard drives

Locate photographs – GPS linking

- Field photographers should always collect a GPS track while in the field.
- Be sure to take a clear photograph of the operating GPS screen showing the date and time to synchronize the photographs with the GPS track (see photograph). The ideal GPS photograph should clearly show, with no clear, the GPS clock in Hours, Minutes, and Seconds
- With the synch photo and a track file, all photographs can be linked to a specific Lat/Long/Time using special software (GSP-Photo Link or OziPhotoTool). However, a different team would likely be responsible for processing GPS-camera information. They

will the synch photo and downloaded track file (using the software that came with the GPS) to GPS-link these photographs.

Photograph logging

- Locating photographs in space and time is a good first step to ensure that photographs become data and not useless files. This can be achieved by creating photograph logs.
- A log can be a simple spreadsheet that captures basic information about each photograph. It can also be a photo database that stores more information and provides additional functionality. A photograph log should include:
  - Photographer name
  - Date
  - Case/incident
  - Note/Caption

Process: Image analysis

- Software like SigmaScan Pro can be used to process photographs. Photo analysis applications can quantify area, percent cover, counts of objects, etc. and it is usually faster and more accurate than manual methods. Consider this if you plan to obtain quantitative data from photographs.

Appendix A
Supporting Documentation – Photography Checklist and Forms

Photography forms may be provided by the NRDA lead, otherwise, use the attached form.

- Print the form on weather-resistant paper (if available). Make more than enough copies of the form before going into the field.
- Fill out forms with waterproof pen or permanent marker. Do not use pencil or biro (erasable) ink.
- Fill in blanks with "N/A" if data are not applicable or not available. Avoid leaving blank values on data forms.
- Do not erase or black out erroneous entries on the field data forms. Errors should be corrected by crossing out the entry with a single line and signing and dating the strike-through.

Attached form:
- Photography Complete Gear Checklist
- Photograph and GPS Checklist
- PhotoLogger Form

Photography Gear Checklist

Photography - Complete gear list; F indicates field gear		
☐	Camera ***F***	With neck strap
☐	Camera case ***F***	Sized to hold all camera gear; plus polypropylene freezer bag, if appropriate
☐	Memory cards ***F***	1-2 extra depending on size – (e.g., 200-300 high resolution photographs, each)
☐	Rechargeable batteries ***F***	Camera batteries: 2 is OK, 3 is better. AA's two sets of rechargeable are OK – extra alkaline or lithium for GPS etc.
☐	Battery charger	Appropriate for each type of rechargeable batteries
☐	Lens cleaning kit ***F***	(e.g., soft cloth)
☐	Card reader	One that accepts many types of cards is preferred
☐	Cable – Camera to PC	
☐	Camera manual	Paper and pdf
☐	Underwater housing/kit ***F***	Optional - useful in rough weather and small boat ops.
☐	Photo scale ***F***	15 cm waterproof, 15 cm disposable. Avoid white or light colors. Grey is best
☐	Image viewing software	All PC's and many cameras have software for reviewing photographs
☐	Image editing software	Optional. Good for processing photographs for presentations etc.
☐	External hard drives	
☐	PhotoLogger database	
☐	GPS-Photo Link software	
☐	DVD-R's – NOT RW's	
☐	Waterproof bag ***F***	Dry sack or heavy duty zip-lock bags
☐	Polarizing lens ***F***	Optional – reduces glare and reflections
☐	GPS ***F***	
☐	Field notebook ***F***	

Photograph and GPS Checklist

The following checklist will help ensure that ALL NRDA photographs and GPS photographs are successfully processed and included in the larger photography database.

Pre-Field:

- ☐ GPS/camera must be set to local time (Set to 24-hour Military Time)
  Garmin GPS: Turn GPS Unit ON> Menu > Setup > Time > Time Format = "24 hours"
- ☐ Datum = "WGS 84"
  Turn On GPS > Menu > Setup > Units > Map Datum = "WGS 84"
- ☐ Set Track Log to "Wrap When Full"
  Turn On GPS > Menu > Tracks > Track Log (Track Log = "On") > Setup > Check "Wrap When Full"
- ☐ Set GPS to Decimal Degrees
  Garmin GPS: Menu> Menu > Setup> Units > Position Format > hdd.ddddd

In-Field:

- ☐ Ensure GPS unit has acquired a satellite signal
  The device may momentarily lose its signal if you are traveling quickly, if you have placed it in an obstructed location (backpack, field kit, etc.), or it is not facing upwards in a secure location. Attaching the GPS to the outside of your pack can be helpful
- ☐ Take a photograph of the GPS unit showing the Date and current Time of day (with seconds) – NOT a waypoint time. See examples below
  At the beginning of the day, take a photograph of the GPS unit with the display screen showing the current date and time (with seconds). DO NOT take a photograph showing the time that a waypoint was taken since this is not the real-time GPS time. To display date/time on Garmin units, press the "Menu" button twice. Make sure the screen is clearly visible and then take a photo (double-checking the photo to make sure the information is captured)
- ☐ Do not delete any photographs in the field!
  Cameras typically auto-number photographs – any gaps in the number sequence may suggest that the camera was tampered with, rising legal defensibility concerns
- ☐ Do not open photographs before zipping – may change metadata
  Original photographs MUST NOT be opened at any time (beyond viewing them on the camera's LCD screen). Only copies may be viewed. Opening the photographs prior to uploading to the NRDA site changes the Date/Time in which the photograph was "modified." This suggests that the photo collection may have been tampered with, thus potentially rendering the collection indefensible
- ☐ Take informative photographs that tell the story
  Photographs of the GPS unit after the beginning of the trip, compass settings, trip preparation, equipment cleansing, sediment mixing, and other sample preparation procedures are helpful, but do not need to be photographed extensively. If such photographs are taken they do not require photograph-specific comments in the Photologger form
- ☐ DO NOT turn the GPS unit off at anytime
  DO NOT turn off the GPS unit at any time during the day, even during rest periods. This causes a break in the track log and leads to difficulties in processing the photographs

Post-Field:

- ☐ Extract tracks and waypoints from the GPS unit
  Tracks and Waypoints are stored in the GPS unit. These are requisite inputs for the photo processing software.
- ☐ Connect the GPS unit to a computer (remember your cables)
- ☐ Start Garmin MapSource (or similar)
  1) Click on the "Receive from Device" icon.
  2) Click Find Device (the name of the GPS unit will appear)
  3) Under "What To Receive" > Click only Waypoints and Tracks
  4) Click "Receive" (you will now see the tracks/waypoints for that day)
- ☐ Save .gdb and .gpx files
  1) > Save As… > "YYYY_MMDD_LastName_FirstName" (Save as type: .gdb)
  2) File > Save As… > "YYYY_MMDD_LastName_FirstName" (Save as type: .gpx)
- ☐ Complete the PhotoLogger Form (see below)

- Identify key photographs
  In the photograph-specific comments section, write the photograph name or number for each key picture and “key photograph” as the comment. Key photographs are those which document the effects of the spill and aid in the NRDA process. These include pictures of: samples, tarballs, oil sheens/slicks, oiled vegetation, oiled wildlife, etc. If several pictures are taken of the same oiling observation, pick the best one and mark it a key photograph. Non-key photographs are those taken of the GPS unit showing the time and date at the beginning of the trip (this should be the 1st photograph in the set), directional photographs (N, E, S, W), and the background landscape (unless it is covered in oil). **Non-key photographs do not require photograph-specific comments**
- Process photographs using GeoJot, see GeoJot training document
- Import photographs to desktop Photologger
- Upload photographs to on-line Photologger
- Upload photograph to ERMA

PhotoLogger Form

This form MUST BE filled out to accompany all NRDA photographs taken in the field. Fill out one form per location or sampling mission

Photographer: _____
Sampler:_____
Workgroup:_____
Work plan:_____
Agency / Cell:_____
Date of photographs: _____
Photo Range *(if submitting multiple forms)***: _____**
Time Zone: AST/ADT
Location where photographs were taken - *Describe the geographic location where the field work was completed*

General description of all photographs

Keywords – that describe all photographs being submitted - *Specific keywords that describe ALL the photographs this form addresses. If you choose to fill out the next section or review photographs in the PhotoLogger database, add keywords for each unique photograph.*

Enter photograph-specific comments here (optional) - *Use this section to call attention to specific photographs of high value. Provide specific comments / keywords. Use this section to identify specific photographs of sample locations/sites, or photographs that are data themselves (e.g., photo plots).*

Photograph Name	Comment and Keywords[1]

[1]Suggested Keywords

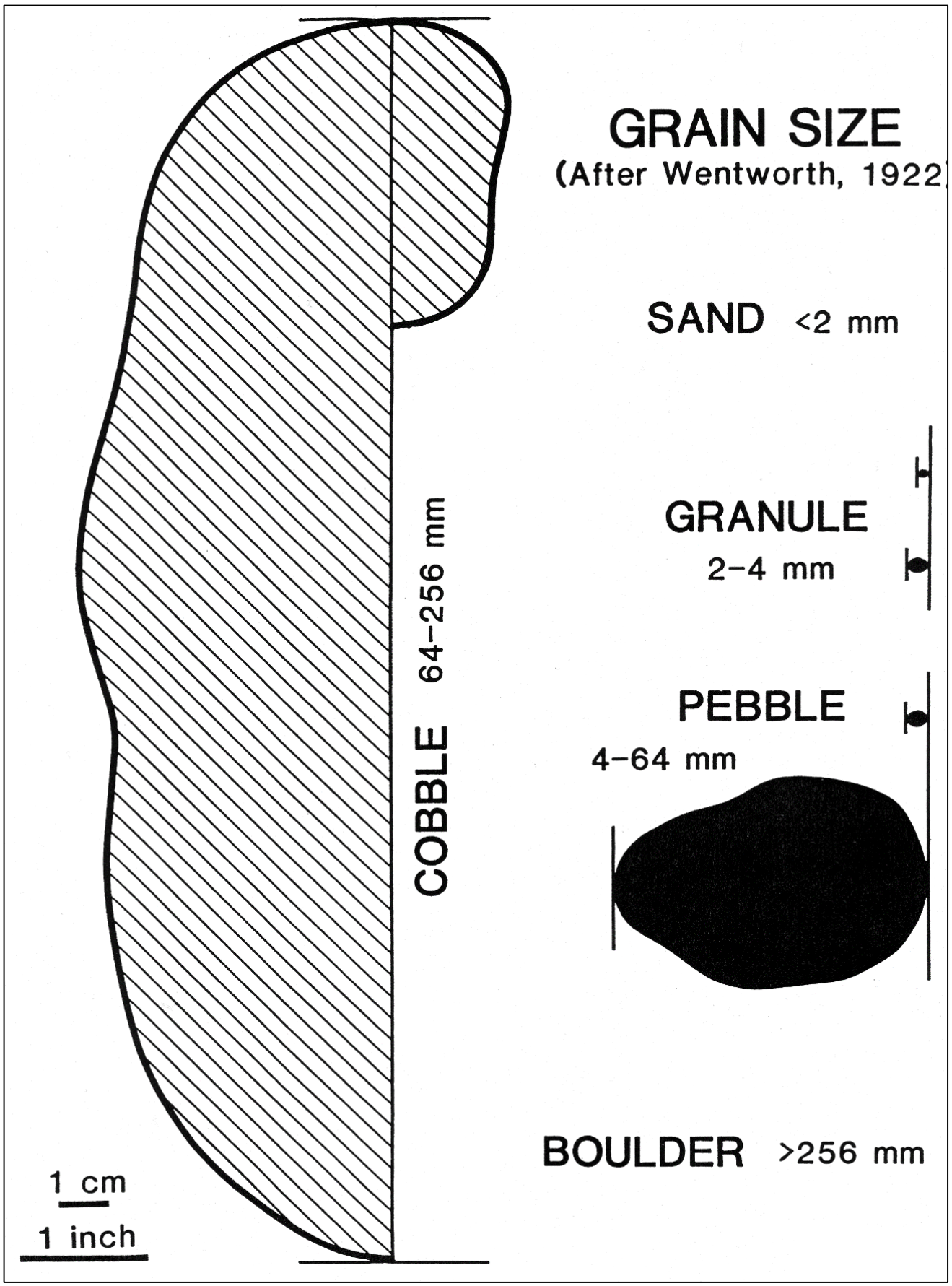
Barge
Barrel
Beach
Birds
Boat
Boom
Container
Chemical
Cleanup Operations
Dead Wildlife
Dispersant
Dredging
Drilling Platforms
Eelgrass
Fish
Fish Kill
Gravel Beach
GPS Unit
Grounding
Ice
Intertidal
In-situ Burning
Invertebrates
Jar
Kelp-Boulder Field
Lagoon
Managed Area
Marine Debris
Marine Mammal
Marsh
Mudflat
NRDA
Oil-Sheen/Rainbow
Oil-Dark
Oil-Emulsified
Oil-Tarball
Oil-Tarmat/Patty
Oil-Surface Residue
Oil-Stain/Coat
Overflight
Pipeline
Pits and Trenches
Quadrat
Response Vessel
Recreational Area
Riprap
Rocky Shore
Sampling
SCAT
Sediment core
Shellfish
Shoreline
Small Boat
Snow
Source Oil
Subtidal
Sunken Vessel
Tank
Tanker/Ship
Transect
Tundra
Waste Site
Wetland
Whale
Wildlife

OTHER SUPPORTING MATERIALS

GRAIN SIZE CHART

General Class	Wentworth Scale (Size Description)		Phi Units ø*	Grain Diameter d (mm)
GRAVEL	Boulder		-8	256.0
	Cobble		-6	64.0
	Pebble		-2	4.0
	Granule		-1	2.0
SAND	Sand	Very Coarse	0	1.0
		Coarse	1	0.5
		Medium	2	0.25
		Fine	3	0.125
		Very Fine	4	0.0625
MUD	Silt		8	0.00391
	Clay		12	0.00024
	Colloid			

* $\text{ø} = -\log_2 d$ (mm)


GRAIN SIZE
(After Wentworth, 1922)
SAND <2 mm
GRANULE
2–4 mm
PEBBLE
4–64 mm
COBBLE 64–256 mm
BOULDER >256 mm
1 cm
1 inch

Disclaimer

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