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

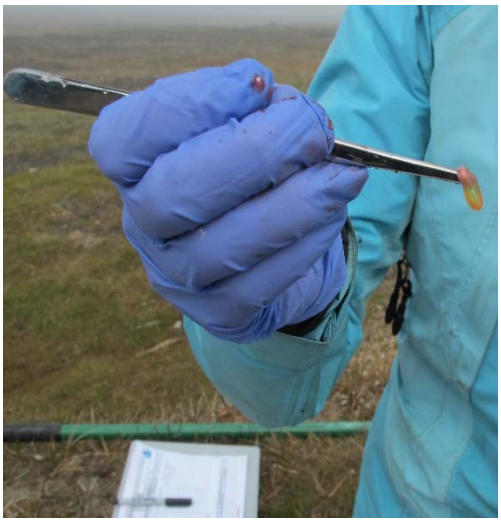
observations or evidence of scavenging. Consider collecting samples for to determine cause of death or for chemical analysis.

- 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)			