14.0 PROTOCOL FOR SAMPLING MUSSELS FOR METALS AND TRACE ORGANICS

Overview

This protocol details methodology to collect indigenous species of mussels. If possible have these shipped to a laboratory for analyses or for transplanting in other areas for a specific time period.

Sources

Gulf of Maine Council (1992), Gulf of Maine Council (1992)

At a glance

Collecting indigenous mussels

1 Collect mussels from a sub-tidal (below the low water line) or low intertidal segment of the shoreline.

2 Ensure that at least one collection is timed to avoid unusual sediment re-suspension by storms or stormwater runoff.

3 Collect mussels in replicate in order to examine within site variation.

4 Collect mussels from each of four discrete areas of the sub-tidal zone (total n=200). The replicates areas should be within a 50 m section of the shore so that they represent one zone of water quality and environmental conditions.

50-60 mm

5 Ensure that all indigenous mussels are of a standard 50 to 60 mm shell length.

6 Wash all mussels to remove soft external growth, sediment, and debris using clean seawater from the collection site. Avoid damaging the byssus as this affects growth and survival of the mussel.

7 Place the mussels on a bed of seaweed or in clean containers (e.g. gallon size wide mouth glass jars) with cool packs, and transport them to the laboratory.

8 Use these mussels for analysis or for subsequent transplanting to other sites.

Transplanting mussels

1 Use four cages, each containing 50 mussels, at each site when transplanting mussels for a 60-day deployment period.

2 Mark 15 of the 50 mussels to be placed in each cage on the left posterior side of the shell using a high speed engraving tool ("Dremel"-type**).** Take care to mark the shell deep enough to be able to read the numbers on retrieval but not so deep as to penetrate the shell and injure or kill the animal.

3 Measure each numbered mussel and record the shell length to the nearest 0.1 mm using a vernier or digitaI caliper so that growth over the deployment period can be determined.

4 Place each group of mussels into separate clean containers after marking and sorting the mussels into replicate groups and hold them in a refrigerator (or a cooler with freeze packs) until

deployed into their cages (23 mm x 23 mm x 23 mm polypropylene moulded baskets). Ensure that mussels are not held out of water for more than two days.

5 Secure the lids with nylon "pull ties". Fasten the cages together with the pull ties and run the mooring line through the cages. Attach any instrumentation, such as the recording thermometers to the outside of the cage

6 Suspend the cages in the water column by means of a subsurface float (e.g. an 8" Styrofoam trawl float). Arrange the mooring gear so that cages are suspended 1 m off the bottom but at a depth that ensures that the cages are underwater at low tide. In areas with a strong current, polypropylene encased steel cable can be used for mooring lines. In addition, either 1 or 2 concrete blocks may be used as inexpensive moorings.

use adequate mooring in strong currents

7 Take precautions to prevent chafing, which can sever the mooring line from the blocks. If the current is strong, use multiple moorings holding fewer cages.

8 Check cages every 2-4 weeks depending on the site conditions. Clean cages of all fouling that would interfere with sea water exchange. Inspect all lines, fastenings and mooring blocks for chafing. Where required re-position, repair and adjust cages if necessary.

9 Retrieve mussels after the period established in the study design (typically 60 days). Clean all mussels from each of the four cages of external debris and rinse them in seawater from the site. Place the mussels from each cage in clean, clearly labeled glass containers with aluminum foil over the mouth of the jar for transport to the laboratory in coolers with freeze packs.