FIELD AND LABORATORY TECHNIQUES GUIDE FOR MUSSEL GROWERS' WATER QUALITY MONITORING PROGRAMME*

FIRST EDITION

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1. INTRODUCTION

This manual is intended to serve as a field and laboratory guide for use by mussel growers in collecting and processing water quality samples at their growing sites. The manual should be carried in the field whenever data and samples are being collected, and it should be referred to when processing samples in the laboratory.

The manual is intentionally brief, presenting the techniques in a cook-book fashion, but with enough detail to allow persons having little or no experience to perform the required tasks as easily and consistently as possible. Another manual, giving details of the rationale behind the different methodologies, as well as information on how data should be interpreted, is scheduled for preparation and distribution in the near future.

No manual, however well-written, is wholly complete. If you require more information, or have comments to offer, please contact the Acadia Centre for Estuarine Research at 902-542-2201, or write to P.O. Box 115, Acadia University, Wolfville, Nova Scotia, B0P 1X0.

2. SELECTION OF SAMPLING SITES

The most important decision that each grower must make at the beginning of this project is where the samples are to be taken. For convenience, we shall distinguish between PRIMARY and SECONDARY sites.

2.1 Primary Sites

These are sites that will be visited on every sampling date, and at which all necessary monitoring activities will be carried out. These sites will provide the basic data set that will be recorded for your growing site. Each grower should choose <u>two</u> primary sites; one at a location containing young mussels (less than one year old) and one at a location containing old mussels (greater than one year old). Once chosen, these sites should be clearly and distinctly marked (e.g., with a numbered float).

There are several important criteria that should be met in selecting the primary sites:

a) The site should be as <u>representative</u> as possible of conditions within your growing site. It should not, for example, be unusually exposed to strong winds, tidal or river currents, or influenced by shoreline activities. A location about 1/3 of the way into a major growing area, where depth is about average for the site, and current flows are also typical, would be a good location for a primary site.
b) Ease of access is important, because the site must be visited even when no other activities are necessary. However, avoid establishing the site in or too near boating channels (including your own normal route if one exists) because oils and other pollutants associated with boating activities may affect water quality and hence mussel growth.

2.2 Secondary Sites

Because water conditions vary to some extent from one part of a growing area to another, and are influenced by the mussels themselves, you may want to consider sampling a number of other locations using the direct recording techniques available to you. This would include salinity, temperature, Secchi depth and dissolved oxygen. If several secondary sites distributed over a growing area are chosen to reflect the variety of depth, current flow and growth potential conditions that you think exist, and are visited frequently, it will be possible for you to compare conditions throughout your growing area with the more detailed information being provided for the primary sites. You may also want to use these techniques to examine sites that you may be considering as future growing sites.

2.3 Site Identification

It is important that all data sheets and samples be clearly identified as to the sites from which they originate; this is especially true of data and samples that will be sent to ACER for

analyses. To avoid confusion we will adopt the following procedure for identification of data sheets and samples:

a) Each grower will be given a number between 1 and 20; this will be referred to as the grower's number.
b) The two Primary sites will be distinguished by an additional number; we suggest that in the beginning you use 1 for the site containing young mussels and 2 for the site containing older mussels.
c) Secondary sites should be distinguished by an additional letter rather than a number.

So, if your grower number is 10, data sheets and samples from your Primary sites should be labelled 10-1 and 10-2, and secondary sites 10-A, 10-B, 10-C, etc.

3. COLLECTING AND RECORDING DATA

3.1 Important Points

The following are a few important points to keep in mind when collecting and recording data and performing laboratory tasks:

a) Unless you like to SCUBA dive, use safety lines on all equipment being used in the field.
b) Be sure to use a waterproof marking pen when recording data and/or labelling samples (a lead pencil is preferable for paper, a grease pencil for glass and plastic).
c) Do not rely on your memory to record data/information at a later time. Details of where, when and how samples are collected are easily forgotten or confused and should always be recorded immediately.
d) Treat your data records as though they were money:
   - store in a safe place,
   - keep duplicate records for your own files,
   - never destroy raw data or field notes - even if they have been recopied.
e) Handle all chemicals with EXTREME CARE. Not only are they very valuable (you cannot carry out the tests without them), but some are hazardous:
   - Sulphuric acid and Lugol's Iodine Solution are caustic,
   - Lugol's Iodine Solution, Reagent No. 2 and Formaldehyde are potentially carcinogenic,
   - most are toxic.

In all cases, avoid breathing fumes, and wash off immediately if some is spilled onto the skin.

3.2 Sampling Depths

A number of the procedures described in this manual require that you take a water sample at a specific depth. This applies to phytoplankton, chlorophyll, particulate organic and inorganic matter and suspended particulate matter. This raises the question as to what depth water samples should be taken. Since one of the goals of the monitoring programme is to relate the growth and condition of mussels to environmental factors, water samples should be taken at a depth corresponding to the depth at which your mussels will be sampled.

For reasons which will be described later (Section 6.11), mussels will be sampled at about the middle of the sock and your water samples should be taken at the corresponding depth. If this depth changes over time, as a result of either your activities or the natural increase in weight as the mussels grow, your sampling depth should change accordingly.

In the case of water samples for dissolved oxygen, you may want to sample at more than one depth, especially if the water column is stratified (i.e., strong differences in temperature and/or salinity with depth). In this instance water samples should be taken at the

surface, in the pycnocline and at the bottom.

3.3 Sample MASTER DATA SHEET

The following page contains a sample of the MASTER DATA SHEET that should be used to record the information that will be sent to ACER for analysis. It should be completed at the time samples are being collected and/or analyzed. The majority of information requested is self-explanatory, but some requires familiarity with the procedures described in Section 6 of this manual.

MASTER DATA SHEET

Mussel Growers' Water Quality Monitoring Programme
Acadia Centre for Estuarine Research

Name _____ Date _____ Time _____ A.M. P.M. Site No. _____

Tide State _____ Water Depth _____ m

Weather Conditions: Cloud Cover _____%
Wind Direction: N NE E SE S SW W NW
Precipitation: Clear Fog Rain Snow Other _____

Sea Conditions: Smooth Rippled Swells Chop Cresting Waves

Water Color _____ Secchi Depth: Disappeared _____ m Reappeared _____ m

Depth of Mussel Lines: Top _____ m Bottom _____ m

Water Chemistry: Salinometer Calibration _____ °/oo

Depth (m)	Temp (°C)	Sal (°/oo)	σ_t	DO B#	DO mg/l	°/oo SAT	CHLOR B#	CHLOR ug/l	TPIM B#	TPIM mg/l	TPOM mg/l	SPM B#	SPM mg/l

Spat Collector _____ Removed: __ yes __ no; Replaced: __ yes __ no

Water Current Clod _____ m Removed: __ yes __ no; Replaced: __ yes __ no

Observations:

a. Mortality

b. Predators

c. Fouling Organisms

d. Miscellaneous

3.4 Sample Label for Zooplankton, Spat, Clod and Mussel Samples

Mussel Growers' Monitoring Programme
Acadia Centre for Estuarine Research

Site # _____ Date _____

Type of Sample:
Mussels: Age _____
Spat Collector: Date In _____
Date Out _____
Clod: Date In _____
Date Out _____
Zooplankton: Depth of Haul _____

4. EQUIPMENT LIST

4.1 Field Equipment

Y-S-I Salinometer and 15 m Probe
Water Sampler w/ Messenger and 10 m Line
Secchi Disc w/ 10 m Line
Zooplankton Net w/ 10 m Line
6 BOD bottles
250 ml Reagent 1 Bottle w/ Dropper
250 ml Reagent 2 Bottle w/ Dropper
6 1-liter Polyethylene Sample Bottles
4 Mason Jars
2 Phytoplankton Vials
Squeeze Bottle
Water Movement Clods
Spat Sample Collectors
Plastic Bags
Clipboard
Data Sheets
Labels (for zooplankton, spat, current clod and mussel samples)
Marking Pens/Pencils

4.2 Laboratory Equipment

Dissolved Oxygen Apparatus
- Burette Stand
- Burette Clamp
- 50 ml Titration Burette
- Funnel
- 250 ml Titration Flask
- 250 ml Graduated Cylinder

Filtering Apparatus
- Styrofoam Place Mat to hold Filtration Apparatus
- Hand-operated Vacuum Pump w/ Gauge and Water Trap
- Magnetic Filter Holder
- Vacuum Filtration Flask
- 1000 ml Graduated Cylinder
- Forceps
- Watmann GF/C Glass Filters (unweighed)
- Watmann GF/C Glass Filters (weighed in containers)
- Millipore 0.45 um HA Filters
- Filter Containers

Reagents
- 500 ml Manganous Sulphate Solution (DO Reagent #1)
- 500 ml Sodium Azide/Sodium Iodide Solution (DO Reagent #2)
- 500 ml Concentrated Sulphuric Acid
- 1000 ml PAO
- 100 ml Saturated Starch Solution
- 50 ml Lugol's Iodine
- 1000 ml Formaldehyde
- Salinometer Calibration Solutions
- Cleaning Solution for Salinometer Probe

5. SEQUENCE FOR FIELD AND LABORATORY TASKS

The following is a suggested sequence for performing the field and laboratory tasks. You may want to amend it to suit your particular preferences

5.1 Field Tasks

a) Prior to departure to sampling site:
   - check field equipment list (Section 4.1) against equipment in field box
   - calibrate salinometer and record reading on MASTER DATA SHEET
b) Locate sampling station
c) Make observations on:
   - tide state, water depth, weather conditions, sea conditions, water color
d) Take a Secchi depth reading
e) Determine if water column is stratified (i.e., do a temperature/ salinity profile)
f) Take water samples and record depths and bottle numbers for:
   - dissolved oxygen (fix if you plan not to return to your lab within 1 hr.)
   - SPM
   - TPIM and TPOM
   - Chlorophyll
   - Phytoplankton
g) Do a zooplankton vertical haul
h) Remove and replace spat collectors
i) Remove and replace water current clod
j) Sample the mussel population
k) Make observations on mortality, predators, fouling organisms, and other items that may be of interest

5.2 Laboratory Tasks

a) Fix dissolved oxygen samples (if not previously done in field)
b) Preserve spat collector samples
c) Preserve zooplankton samples
d) Preserve phytoplankton samples
e) Preserve spat samples
f) Filter chlorophyll, TPM and SPM samples
g) Do dissolved oxygen titrations
h) Freeze filters, mussel and spat samples

6. TECHNICAL PROCEDURES

6.1 Secchi Disc Readings

The Secchi disc is used to obtain a crude estimate of the depth to which sunlight penetrates the water. Its use is simple:

a) Slowly lower the disc into the water and note the depth at which it just disappears.
b) Slowly raise the disc and note the depth at which it just reappears.
c) Record both depths on the MASTER DATA SHEET.

NOTE: Readings should be made from the shaded side of the boat to reduce the confounding effect of surface reflection. Estimate the depths to the nearest tenth of a meter.

6.2 Salinity/Temperature Measurements

6.2.1 Meter Setup:

a) Adjust meter zero (if necessary) by turning screw on meter face so that needle coincides with zero on conductivity scale.
b) Turn the MODE control to REDLINE and adjust the REDLINE control so the needle lines up with the redline on the meter face (if this cannot be done the batteries will have to be replaced--refer to the owner's manual for instructions and recommended battery type).
c) Plug the probe into the probe jack on the side of the instrument.

6.2.2 Meter Calibration:

a) Rinse the probe in calibration solutions #1 and #2 in that order.
b) Place the probe in calibration solution #3.
c) Set the MODE control to TEMPERATURE and note the temperature of the calibration solution (you should swirl the probe gently and allow a few seconds for the probe temperature to come to equilibrium with that of the test solution before reading).
d) Adjust the TEMPERATURE CONTROL KNOB to the temperature of the solution.
e) Switch the MODE control to the SALINITY position and read salinity on the red 0-40 °/oo meter range.
f) Depress the CELL TEST button--the meter reading should fall less than 2 °/oo (if greater, the probe is fouled and should be cleaned--see the owner's manual for instructions).
g) Record the salinity of calibration solution #3 on the MASTER DATA SHEET.

6.2.3 Salinity/Temperature Profiles

To obtain depth profiles of salinity and temperature at your sampling sites simply repeat steps c, d and e of the calibration procedure at one meter depth intervals beginning at the surface.

NOTE: Both temperature and salinity readings should be estimated to one decimal place.

6.3 Chlorophyll

6.3.1 Field Procedure

Collect water sample(s) at the desired depth using the water sampler and transfer to 1 liter polyethylene bottles. Be sure to record the appropriate depth and corresponding bottle numbers on the MASTER DATA SHEET.

6.3.2 Laboratory Procedure

a) Using a blunt forceps, place a Watmann GF/C filter on the center of the filter base (a ridge around the base will guide the filter into position).
b) Place the funnel housing on top of the base (a magnet holds it in place).
c) Pour ca. 200 ml of the sample into the filter housing.
d) Turn trigger on bottom of vacuum pump counterclockwise against its stop and create a suction no greater than 20 cm Hg (read on inner scale of vacuum gauge). (See NOTE 1.)
e) Continue to add sample to filter housing until entire sample is filtered being careful not to suck the filter dry until all the sample is processed (see NOTE 2).
f) Release suction (turn trigger clockwise) immediately after the last of the sample goes through the filter.
g) Remove funnel housing and, using blunt forceps, remove filter, place into a container and cover. (See NOTE 3.)
h) Label sample for SITE #, DATE and ML FILTERED (the latter is best estimated by transferring the filtrate into a 1000 ml measuring cylinder).
i) Freeze sample as soon as possible.

NOTE 1: Too great a suction will break down the plant cells and allow chlorophyll molecules to pass through the filter and into the filtrate.

NOTE 2: Sucking the filter dry before the entire sample is filtered changes the retention capacity of the filter.

NOTE 3: Do not touch the filter with your bare fingers. Chlorophyll is a very sensitive molecule and will break down upon contact with the acids in your skin.

Additional NOTE: Since chlorophyll breaks down when exposed to strong sunlight, try to carry out the filtration in subdued light and store samples in the dark.

6.4 Total Particulate Inorganic Matter (TPIM) and Total Particulate Organic Matter (TPOM)

The same water sample is used for determination of total particulate inorganic matter (TPIM) and total particulate organic matter (TPOM).

The field and laboratory procedures for taking and processing TPIM and TPOM samples are the same as for processing chlorophyll samples except that a pre-weighed, pre-combusted Watmann GF/C filter is used. These are supplied in individual filter containers having the filter's weight recorded on the label (therefore, be careful not to switch filters to different containers). In addition, care should be taken not to contaminate the sample with foreign particles such as wood and paint chips from the boat, air-borne dust particles in the laboratory, etc.

6.5 Suspended Particulate Matter (SPM)

The procedure for collecting and processing SPM samples is the same as for chlorophyll and POC except that a 0.45 um HA Millipore filter is used. This type of filter requires a bit more attention in use:

a) Unlike the Whatmann GF/C filters, these have a top and bottom and you should take care that the filter is placed top up on the filter base. The top is the side that is presented to you as you remove the filter from its package.
b) A blue paper separates the filters--be sure this separator is not carried along with the filter and placed on the filter base.
c) If there is a large amount of suspended material in the sample, you may not be able to filter the entire 1000 ml. In this case the best procedure is to add 200 ml aliquots at a time and wait until each aliquot is almost completely filtered before adding more. If you find that the filtration rate is decreasing rapidly, do not try to filter the entire sample, but do try to filter as much as possible. (In these instances, it is permissible to increase the suction to ca. 40 mm Hg--but only for SPM.)

6.6 Dissolved Oxygen

6.6.1 Field Procedure

a) Using the water sampler, collect a water sample from the desired depth.
b) Transfer the water sample to a BOD bottle with as little agitation as possible using the following technique:
   - insert the siphon of the water sampler into the bottom of the BOD bottle.
   - open the spigot of the water sampler and allow the BOD bottle to overflow 2 to 3 times its volume.
   - slowly withdraw the siphon without closing the spigot.
c) Fix the sample by adding 2 ml of Reagent #1 followed by 2 ml of Reagent #2.
d) Incline the BOD bottle slightly and, with a quick twisting motion, introduce the glass stopper. Be careful not to trap air bubbles in the BOD bottle at this point--if this happens you will have to discard the sample.
e) Mix the reagents with the sample by rapidly inverting the BOD bottle 10-20 times using a strong wrist-snapping motion. (If dissolved oxygen is present a brownish floc will form.)

6.6.2 Laboratory Procedure

a) Using a funnel, fill the titration burette with PAO titrant (see NOTE 1).
b) Check to be sure that the floc in the BOD bottle has settled so the upper one-third of the bottle is clear. If not, allow more time for the floc to settle.
c) Once the floc has settled remove the stopper, add 2 ml of concentrated sulphuric acid, restopper and mix using the same technique as in step d and e of the field procedure.
d) Using the 250 ml graduated cylinder, transfer 200 ml of the sample to a titration flask.
e) Add 5-10 drops of starch solution and gently swirl to mix.
f) Note the ml of titrant in the burette and record this on the DO data sheet (see NOTE 2).
g) While gently swirling the flask, slowly add titrant to the sample until the blue color just disappears. Disregard any reappearance of the blue color (see NOTE 3).
h) Record the ml of titrant remaining in the burette on the DO data sheet.
i) Calculate the mg/l of oxygen in the sample as follows:

$$\text{mg/l DO} = \frac{200 \times \text{ml titrant used}}{\text{volume titrated in ml}}$$

j) Calculate the percent saturation as follows:

Using Rawson's nomogram on the bottom of the DO data sheet, place a straight edge to line up with oxygen concentration on the lower scale and with water temperature on the upper scale. Percent saturation is read off where the straight edge crosses the middle line.

k) Record mg/l DO and % saturation on the MASTER DATA SHEET.

NOTE 1: a) Be careful that you do not trap air bubbles in the burette or this will result in inaccurate readings. If this does happen the bubbles can be removed by either tapping the side of the burette or, with your finger over the top, inverting the burette several times.
b) Be sure to fill the tip of the burette by allowing a small amount of titrant to run out the stop-cock.

NOTE 2: It may be helpful to place a piece of white paper behind the burette to make it easier to read.

NOTE 3: If you accidentally overrun the end-point of the titration, add an additional 20 ml of your fixed sample to the titration flask. This should make the color reappear. Re-titrate, remembering to correct for the volume titrated when using the formula in instruction i above.

Additional NOTE: If you have PAO titrant remaining in the burette after completing the titrations, it should be discarded--<u>DO NOT</u> pour it back into the PAO reagent bottle.

6.6.3 Sample Calculations

a) Using 100 ml and titrating accurately to the endpoint using 3.0 ml of PAO:

$$\text{Dissolved Oxygen (mg/l)} = \frac{200 \times 3.0}{100} = 6.0$$

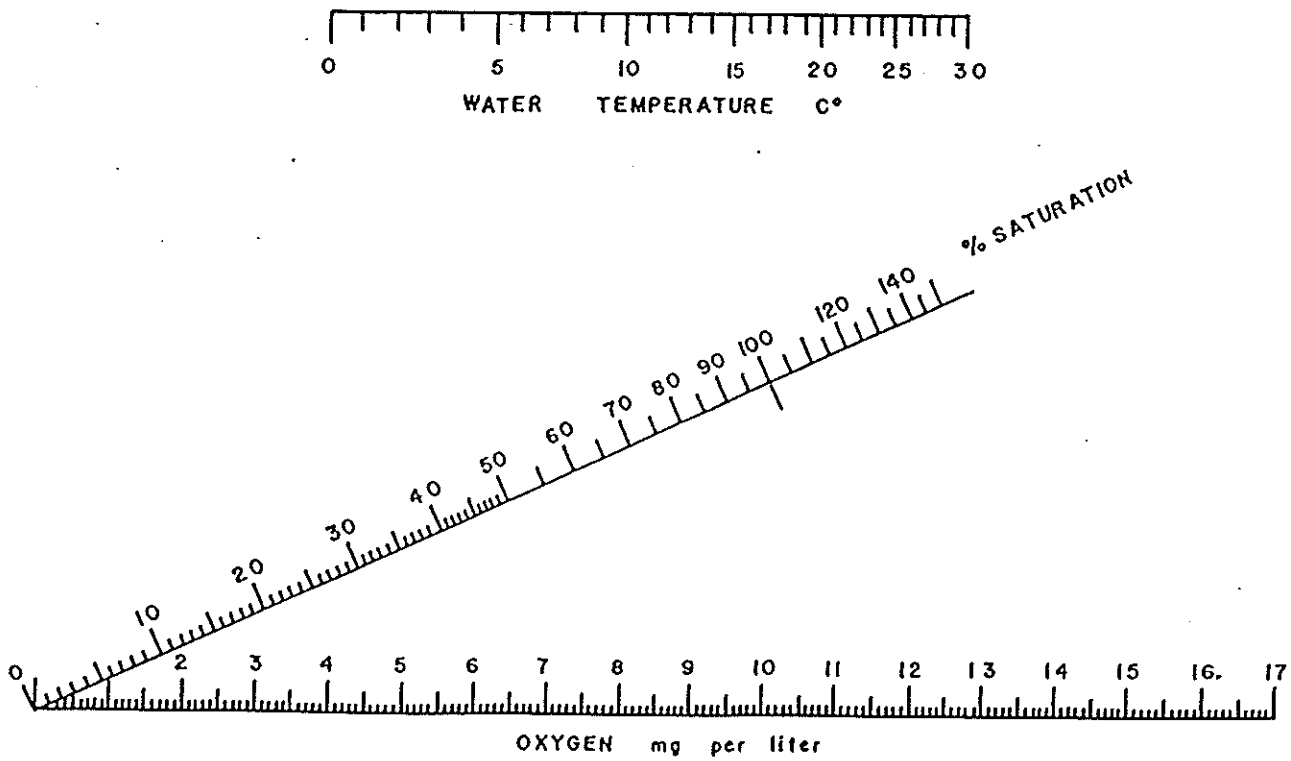
b) Overshooting the endpoint, adding an additional 20 ml of fixed sample, and then titrating accurately to the endpoint using a total of 3.6 ml:

$$\text{DO (mg/l)} = \frac{200 \times 3.6}{120} = 6.0$$

DISSOLVED OXYGEN DATA SHEET

Name _____ Date _____ Site # _____

Bottle No.	ml titrant Start (A)	ml titrant End (B)	ml titrant used (B-A)	DO (mg/l)	Temp (^{o}C)	Percent Saturation



Rawson's nomogram for calculating percent saturation of dissolved oxygen in water in relation to temperature from saturated air at 760 mm Hg pressure (Welch, 1948).

6.7 Phytoplankton

6.7.1 Field Procedure

Water samples for phytoplankton analysis should be taken with the water sampler. Transfer the sample to a 20 ml vial and mark the vial for SITE # and DATE using a grease pen.

6.7.2 Laboratory Procedure

Add 2-5 drops of Lugol's Iodine and shake. The sample should be the color of weak tea. Store refrigerated and in the dark--keep from freezing.

6.8 Zooplankton

6.8.1 Field Procedure

a) Before using the plankton net, check that it is clean, does not contain remnants of previous samples, and that the plastic clip is closed to prevent loss of the sample.
b) Allow the weighted net to descend to the full length of the 10 meter line, making sure that the net or rope does not become twisted.
c) Leave the net at its maximum depth for a few seconds and observe whether the line is vertical, or whether currents are dragging the net away. Make a note of any strong displacement from the vertical.
d) Retrieve the net by pulling steadily; do not jerk the line or pull it too fast. A slow, steady pull at a speed that will bring the net to the surface in about 20 seconds is about right.
e) Bring the net into the boat and, using seawater from the squeeze bottle, wash the inside of the net so that all the zooplankton are contained within the plastic cup and tube at the bottom.
f) Hold the tube of the net over a 500 ml Mason jar, open the plastic clamp and drain <u>all</u> the contents into the jar. You may have to use a small amount of seawater from the squeeze bottle to fully empty the net.
g) Check to be sure that all contents have been washed out of the net. Reset the clamp (to avoid mistakes on the next use) and store the net away.
h) Record the appropriate data on a label, insert the label into the sample jar and seal the jar.

6.8.2 Laboratory Procedure

a) Add a volume of concentrated Formalin solution to the jar equivalent to about 1/10 the volume of the sample.
b) Store the sample in a cool place, but keep from freezing.

6.9 Spat Collections

Monitoring the time and density of spatfall requires that a small spat collector be placed at a convenient location in your growing area. A variety of materials are available as substrates for collecting spat; we have chosen to use a piece of the small plastic 'scrubbies' commonly available at most grocery stores.

6.9.1 Field Procedure

a) Installation - thread the collector through the 1 meter weighted line and attach to an anchored line.
b) Removal - unthread the collector, place in a sample jar along with a completed label and cover with seawater.

6.9.2 Laboratory Procedure

a) Add a volume of formalin equivalent to about 1/10 the volume of seawater in the sample jar, seal and mix.
b) Store in a cool place, but keep from freezing.

6.10 Water Current Clods

These devices are intended to provide an integrated index of water movement at your sampling site. They are made of a mixture of plaster of paris, latex paint, and water and will dissolve at a rate proportional to temperature and current velocity.

Instalation: Attach the clod to a galvanized eyebolt. Tighten the nuts carefully (overtightening will crack the clod). Tie to the eyebolt one end of a length of line long enough to correspond to about mid-depth of your mussel lines (this will be approximate since depths will vary as current velocity varies). Suspend the clod from an anchored float or line close to your monitoring site. Be sure to allow space for the clod to move with the water currents without becoming entangled with other lines.

Replacement: The clods should last for about one month before replacement is necessary. Over this period they should decrease to about 3/4 or 1/2 of their original size. However in areas where current velocities are high, it may be necessary to replace clods every two weeks. You will have to experiment a bit to determine the appropiate replacement time for your particular situation.

To remove the clod simply remove the eyebolt, place the clod in a plastic bag and label as to SITE #, DATE IN, and DATE OUT.

6.11 Sampling Mussels

In order to monitor growth, fecundity and condition of your mussels and relate these factors to environmental conditions, a random sample of mussels from socks at the Primary sites is required. Unfortunately, opinions differ among biologists as to the proper procedure for sampling mussels. Growth of mussels within one sock is highly variable; those in the interior of a sock may grow poorly, and there may be considerable differences between the top and bottom of a sock. As a result, some biologists feel that only the outer mussels in the middle of a sock should be sampled, while others feel that both inner and outer mussels, from top, middle and bottom of the sock, should be sampled.

What is most important for our purposes is that the sampling technique be consistent. Only if this is the case can meaningful comparisons amoung different times, as well as amoung different sites, be possible. It is therefore imperative that all mussel samples be taken in the same manner.

Our feeling is that a good compromise for a limited sampling programme is to sample both inner and outer mussels at about the middle of the sock. You will, however, have to be careful about the effects sampling will have on growth rates. If you continuously sample from the same sock at the same depth you will probably increase overall growth rates as a result of thinning. To avoid this you should spread your sampling over a number of socks, but all socks should be located near to each other.

To take a sample, remove several handfuls of mussels from the middle of the sock, being sure to collect mussels from both the inner and outer parts of the sock, place in a bucket, mix and randomly remove about 20-25 mussels. Place this sample in a single plastic bag, enclose a completed label and seal.

7. MATERIALS TO BE SHIPPED TO ACER

The following list includes the materials that should be shipped to ACER for analysis:

Chlorophyll Filters
POC Filters
SPM Filters
Zooplankton Samples
Phytoplankton Samples
Spat Collection Samples
Water Movement Clods
Mussel Samples
Copies of MASTER DATA SHEET(S) and BOD DATA SHEET(S)

8. SHIPPING INSTRUCTIONS

Samples should be carefully packed in styrofoam coolers or cardboard boxes, sealed with duct tape, marked "PERISHABLE AND FRAGILE" and sent to:

Acadia Centre for Estuarine Research
P.O. Box 115
Acadia University
Wolfville, Nova Scotia
B0P 1X0

NOTE: Samples should be sent collect to ACER via any local courier service. Be sure to send samples no later than wednesday of the week or else they may arrive at ACER during the weekend when there may be no one to accept delivery.

9. ANALYSES BY ACER

Upon receipt, ACER will analyze your data and samples and return the results to you as soon as possible. You will receive the following information for each Primary site:

a) $_t$ values, an index of stability of stratification and graphs of vertical profiles of salinity, temperature and $_t$ for stratified water columns;
b) Chlorophyll concentration;
c) Total particulate inorganic matter concentration;
d) Total particulate organic matter concentration;
e) Suspended particulate matter concentration;
f) Zooplankton density (expressed as settled volume);
g) Spatfall density;
h) Current velocity index;
i) Growth rate of mussels (expressed as dry weight on weekly, monthly and yearly basis);
j) Fecundity estimates (expressed as % gonad weight on a dry weight basis);
k) Glycogen content (expressed as % of total meat weight on a dry weight basis);
l) Condition index (expressed as % meat weight on a dry weight basis).

In addition to receiving the above information, you will also receive the <u>average</u> value of these variables for all other sites being monitored. This will allow you to compare your site conditions and mussels with those of others.

WATER QUALITY MONITORING PROGRAMME FOR MUSSEL GROWERS
ACADIA CENTRE FOR ESTUARINE RESEARCH

SOME REPRESENTATIVE VALUES OF MONITORING VARIABLES *

Parameter	Low	High	Units**
SALINITY	<10	>34	‰
TEMPERATURE	<0	>20	°C
STABILITY OF STRATIFICATION	0	100	g/cm^3
SECCHI DEPTH	<3	>10	m
OXYGEN SATURATION	<50	>100	%
CHLOROPHYLL A	<2	>6	µg/l
PHAEOPHYTIN	-	-	µg/l
TOTAL PARTICULATE INORGANIC MATTER (TPIM)	<5	>50	mg/l
TOTAL PARTICULATE ORGANIC MATTER (TPOM)	<.05	>1	mg/l
SUSPENDED PARTICULATE MATTER (SPM)	<5	>50	mg/l
ZOOPLANKTON	-	-	mg/sample
SPATFALL DENSITY	-	-	#/collector
CURRENT VELOCITY INDEX	-	-	g lost/day
MONTHLY GROWTH RATE (MEAT)	-	-	mg dry wt/month
MONTHLY GROWTH RATE (SHELL)	-	-	mm/month
FECUNDITY (% GONAD WEIGHT)	-	-	% total meat wt
GLYCOGEN CONTENT	<1	>20	% total meat wt
CONDITION INDEX $\left(\frac{\text{g dry meat weight}}{\text{g dry shell weight}}\right) \times 100$	<5	>15	-

* The range of some parameters is difficult to estimate at present because the techniques employed for measurement are not commmonly used or have not been standardized, and there is little information to base estimates on. As the monitoring programme continues and a data base is developed, high and low estimates for these parameters will be provided.

** units: cm^3 = cubic centimeters; ‰ = parts per thousand; m = meters; g = grams; µg = micrograms; mg = milligrams; l = liters