+++ IF 4 < 3 +++

**Color key for this file**

* Purple text is important information that will be turned into Developer code
* Do not touch the developer code that looks like “**+++INS `${title}`+++**”
* Do not touch the developer code that is green highlighted
* Must be provided before the QAPP is considered complete.

+++ END-IF +++

+++EXEC

determine = (mtype, wtype, name, method) => parameters.some((param) => mtype !== '' ? param.monitoringCategory.toLowerCase() === mtype.toLowerCase() : wtype !== '' ? param.waterType.toLowerCase() === wtype.toLowerCase() : name !== '' ? param.parameter.toLowerCase() === name.toLowerCase() : method !== '' ? param.method.toLowerCase() === method.toLowerCase() : false);

determineConcern = (label) => waterConcerns.some((concern) => concern.label.toLowerCase() === label.toLowerCase());

+++

**Quality Assurance Project Plan**

**for**

**+++INS `${title}`+++**

**Prepared by**

**+++INS `${preparedBy}`+++**

*This Quality Assurance Project Plan was generated by AquaQAPP, a tool managed by Massachusetts Bays National Estuary Partnership and developed with funding from the United States Environmental Protection Agency and the Massachusetts Department of Environmental Protection.*

Date generated: +++INS `${dateGenerated}`+++

**Disclaimer:** This plan was generated by AquaQAPP, a web-based application created by the Massachusetts Bays National Estuary Partnership (MassBays). AquaQAPP generates tailored Quality Assurance Project Plans (QAPPs) for marine and freshwater water quality and benthic monitoring efforts in the Commonwealth of Massachusetts and is intended to assist volunteer monitoring programs in collecting quality-assured data.

This plan does not define, or otherwise limit, the purpose for which organizations may seek to use the plan or apply their data. A goal of the AquaQAPP project, however, is to bring more citizen science data to decision makers, including MassBays, the Massachusetts Department of Environmental Protection (DEP), and the Environmental Protection Agency (EPA).

Use of AquaQAPP is not required. By using AquaQAPP to document and plan for collection of quality assured data, the monitoring program’s QAPP is considered pre-approved by DEP and deemed acceptable by EPA, and agency review is not required prior to sampling. This pre-approval is valid so long as samples are collected and analyzed in strict accordance with the QAPP generated by the application which has not been significantly altered from the original output. “Not significantly altered” means that the user has not made or will not make changes to collection protocols, analytical methods, or other substantive content included in the generated QAPP.

Where new or revised methods, additional parameters, or other substantial changes are included beyond the content generated by AquaQAPP, and a stated objective of the monitoring effort is to submit the resulting data to DEP, DEP requires QAPP review prior to implementation. The modified QAPP can be submitted to:

Suzanne Flint (Suzanne.Flint@mass.gov)

Bureau of Water Resources, Watershed Planning Program

Massachusetts Department of Environmental Protection

8 New Bond Street, Worcester, MA 01606

DEP retains sole discretion as to what extent the agency will use data or information produced or resulting from use of this document.

Monitoring programs funded by EPA or DEP must follow agency requirements for quality assurance, and AquaQAPP may or may not meet those requirements. Please check with the funding agency for guidance in that case.

# Section A. Project Management Elements

## A1. Title and Certification Page

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Plan Title:** | +++INS `${title}`+++ | | | |
| **Name of Organization(s) Implementing Project:** | | | |  |
| **Prepared by:** | | +++INS `${preparedBy}`+++ | | |
| **Effective Dates of Plan:** | | |  | |

+++FOR person IN projectOrganization +++

+++IF $person.primaryContact !== 'X'+++

**+++ INS $person.titlePosition+++:**

|  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- |
| **Name:** | +++INS $person.fullName+++ | |  | **Phone:** | +++INS $person.telephone+++ |
| **Signature:** | |  |  | **Date:** |  |

+++END-IF+++

+++END-FOR person+++

+++FOR person IN projectOrganization +++

+++IF $person.primaryContact === 'X'+++

**Primary Contact:**

|  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- |
| **Name:** | +++INS $person.fullName+++ | |  | **Phone:** | +++INS $person.telephone+++ |
| **Signature:** | |  |  | **Date:** |  |

I +++INS $person.fullName+++ certify that +++INS $person.organization+++ [ ] has [ ] has not made changes to methods, or added monitoring parameters beyond the content generated by AquaQAPP.

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Signature:** |  |  | **Date:** |  |

+++END-IF+++

+++END-FOR person+++

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[Table B5.8. Data Validation Quality Control: Enterococci 124](#_Toc17709293)

[Table B5.9. Field Quality Control: Microcystins 124](#_Toc17709294)

[Table B5.10. Data Validation Quality Control for Microcystins 124](#_Toc17709295)

[Table B6.1. Typical Instrument/Equipment Inspection and Testing Procedures 124](#_Toc17709296)

[Table B7.1. Instrument Calibration Procedures 124](#_Toc17709297)

[Table B9.1. Examples of Secondary Data Providers **Error! Bookmark not defined.**](#_Toc17709298)

[Table B10.1. Data Management, Review, Validation, and Verification Process **Error! Bookmark not defined.**](#_Toc17709299)

## A.3 Distribution List

The following individuals and their respective organizations will hold copies of the approved QAPP:

|  |  |
| --- | --- |
| Project Role | Name, Organization |
| Project Manager |  |
| Monitoring Program Coordinator |  |
| Program Quality Assurance Officer |  |
| Project Field Coordinator |  |
| Project Lab Coordinator |  |
| Data Management Coordinator |  |
| Program Participants |  |

+++FOR person IN projectOrganization +++

+++IF $person.distributionList === 'X'+++

|  |  |  |  |  |  |  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- | --- | --- | --- | --- | --- | --- |
| **Name:** | | +++ **INS $**person.fullName+++ | | | | | | | | | |
| **Title:** | +++ **INS $**person.titlePosition+++ | | | | | | | | | | |
| **Organization** | | | | | +++ **INS $**person.organization+++ | | | | | | |
| **Address:** | | | +++ **INS $**person.address+++ | | | | | | | | |
| **City:** | +++ **INS $**person.city+++ | | | | | | | | | | |
| **State:** | | +++ **INS $**person.state+++ | | | | | |  | **Zip:** | | +++ **INS $**person.zip+++ |
| **Telephone:** | | | | +++ **INS $**person.telephone+++ | |  | **Email:** | | | +++ **INS $**person.email+++ | |

+++END-IF+++

+++END-FOR person +++

## A4 Program Organization and Task Responsibilities

Table A4.1. Project Organization and Responsibilities

|  |  |
| --- | --- |
| Personnel name and title | Responsibilities |
| +++FOR person IN projectOrganization +++ |  |
| +++ **INS $**person.titlePosition+++  +++ **INS $**person.fullName+++ | +++ **INS $**person.responsibilities+++ |
| +++END-FOR person +++ |  |

## A5 Problem Definition/Background

### A5.1 Problem Definition

+++INS `${problemDefinition}`+++

### A5.2 Problem Background

+++INS `${problemBackground}`+++

## A6 Project Description and Timeline

### A6.1 Project Description

+++INS `${projectDescription}`+++

### A6.2 Map(s) of Area, Waterbody, and Sampling Sites

~~A map with sampling locations labeled is included as an attachment. The map includes a legend, scale, and compass direction.~~

|  |  |  |
| --- | --- | --- |
| **Location ID** | **Location Name** | **Latitude/Longitude** |
| +++FOR location IN monitoringLocations +++ |  |  |
| +++ **INS $**location.locationId+++ | +++ **INS $**location.locationName+++ | +++ **INS $**location.locationLat+++ / +++ **INS $**location.locationLong+++ |
| +++END-FOR location +++ |  |  |

### A6.3 Anticipated Schedule

Table A6.1. Program Schedule

| Activity | J | F | M | A | M | J | J | A | S | O | N | D |
| --- | --- | --- | --- | --- | --- | --- | --- | --- | --- | --- | --- | --- |
| +++FOR activity IN projectActivities +++ |  |  |  |  |  |  |  |  |  |  |  |  |
| +++ **INS $**activity.activity+++ | +++ **INS $**activity.january+++ | +++ **INS $**activity.february+++ | +++ **INS $**activity.march+++ | +++ **INS $**activity.april+++ | +++ **INS $**activity.may+++ | +++ **INS $**activity.june+++ | +++ **INS $**activity.july+++ | +++ **INS $**activity.august+++ | +++ **INS $**activity.september+++ | +++ **INS $**activity.october+++ | +++ **INS $**activity.november+++ | +++ **INS $**activity.december+++ |
| +++END-FOR activity +++ |  |  |  |  |  |  |  |  |  |  |  |  |

+++IF determine('Freshwater Water Quality', 'Freshwater' ,'','') === true+++

## A7 Freshwater/Water Quality Data Quality Objectives

Requirements for ensuring that the data are usable for their intended purpose (that is, are of suitable quality) include accuracy, precision, representativeness, comparability, and completeness. When these requirements are met, the final data product is technically defensible. Data elements for this project are discussed in terms of the appropriate characteristics, defined as:

|  |  |
| --- | --- |
| **Accuracy:** | The extent of agreement between a measured value and the true value of interest. |
| **Precision:** | The extent of mutual agreement among independent, similar, or related measurements. |
| **Representativeness:** | The extent to which measurements represent true systems. |
| **Comparability:** | The extent to which data from one study can be compared directly to similar studies. |
| **Completeness:** | The measure of the amount of data acquired versus the amount of data required to fulfill the statistical criteria for the intended use of the data. |

Quality objectives are given in A7.1 and A7.2. Details of how these criteria are met for each component of the monitoring program’s monitoring tasks are presented in Section B5.

#### Accuracy

Laboratory accuracy will be established by following the policy and procedures provided in the laboratory’s QAPP. These generally employ estimates of percent recoveries for known internal standards, matrix spikes and performance evaluation samples, and evaluation of blank contamination.

Depending on the analyte, specific accuracy objectives can be concentration-based (e.g., +/- 0.010 mg/L @ < .05 mg/L and +/- 20% @ > .05 mg/L) or can be defined in terms of recovery percentages (e.g., 80–120% recovery of matrix spike/PE sample) (PE refers to Performance Evaluation sample).

Accuracy for multi-probe measurements will be tested pre-monitoring using standards that bracket the measurement range and post-monitoring checked against standards to determine if probes remained in calibration at the end of the measurement period. A National Institute of Standards and Technology-certified thermometer is used to periodically check thermometer accuracy. Lower limit accuracy for dissolved oxygen (DO) will be checked using a zero DO standard (when and where low DO are expected). The post-sampling checks of each unit ensure that the readings taken during the survey(s) were within QC acceptance limits for each multi-probe analyte.

#### Precision

Precision is a measure of the degree of agreement among repeated measurements and is estimated through sampling and analysis of replicate (e.g., duplicate) samples. For water quality measurements, duplicates for 10% of samples per will be taken on every sampling event, as applicable.

Laboratory precision of lab duplicates will be determined by following the policy and procedures provided in the laboratory’s QAPP. This varies depending on the analyte, but typically involves analysis of same-sample lab duplicates and matrix spike duplicates.

Overall precision objectives using relative percent difference of field duplicate samples vary depending on the parameter and typically range from 10% to 25% relative percent difference.

Precision of the multi-probe measurements can be determined by taking duplicate (via a second placement of the unit) readings at the same station location. This is sometimes performed for lake surveys. Multi-probe precision objectives generally range from 5 to 10% RPD depending on the parameter.

#### Representativeness

Representativeness refers to the extent to which measurements represent true systems. Sampling locations and survey times will be selected to ensure that the samples taken represent typical field conditions at the time and location of sampling. Exceptions occur in the case of stations chosen to evaluate site-specific impacts (or “hot spots”) where sampling locations will be selected to representativeness of distinct conditions. Other factors, such as seasonality and weather conditions, will be recorded and considered when evaluating whether the resulting data are representative of (e.g., wet weather water quality).

#### Comparability

Comparability refers to the extent to which data from one study can be compared directly to similar studies. Standardized sampling and analytical methods, units of reporting, and site selection procedures will be used to ensure comparability of data with others using those same methods. Sample time and date of collection, sample storage and transfer, as well as laboratories and identification specialists used will be documented so that future surveys can produce comparable data by following similar procedures.

#### Completeness

Completeness is expressed as a percentage of the number of valid measurements that should have been collected*.* This project will be considered fully successful if at least 80% of the anticipated number of samples are collected, analyzed, and determined to meet data quality objectives. At the close of the project, the Project Manager will produce a report detailing the number of samples collected, number of valid results, and percent completion (number of valid samples/number of samples) for each parameter.

Completeness refers to the measure of the amount of data acquired versus the amount of data required to fulfill the statistical criteria for the intended use of the data. Completeness is expressed as a percentage of the number of valid measurements that should have been collected*.* For this project, at least 80% of the anticipated number of samples will be collected, analyzed and determined to meet data quality objectives for the project to be considered fully successful. At the close of the project, the Project Manager will produce a report detailing the number of samples collected, number of valid results and percent completion (number of valid samples/number of samples) for each parameter

Table A7.1. Data Quality Indicators, Quality Control Activities, and Goals

| Data Quality Indicators | Quality Control Actions and Checks | Typical DQI goals |
| --- | --- | --- |
| **Precision** | Field and laboratory replicates | 20% RPD (relative percent deviation) or RSD (relative standard deviation) |
| **Accuracy** | Calibration standards, blanks | No blanks contaminated and all calibrations within acceptable limits |
| **Representativeness** | Evaluate whether the data accurately represent the system population, places, time and/or situation of interest | Data collected represent the system characterized or exposure experienced and are not biased. |
| **Comparability** | Compare to existing data or datasets | Data collected are sufficiently similar in methodology to permit a meaningful analysis |
| **Completeness** | Compare to intended sampling goals to meet project purpose | Could be stated as total number of samples or % of samples collected (e.g. 90%) or an identification of the critical samples needed for the project purposes. |
| **Sensitivity** | Compare to reporting or detection limits from existing data or for decision-making but generally reporting or detection limit should be 3 to 5 times lower than an action level | State sensitivity needed for instruments, methods or processes used for project to obtain meaningful data. This depends on analytical method |

Table A7.2. Data Quality Objectives for Freshwater Water Quality

| Parameter | Units | Accuracy 1 | Overall Precision 2 (RPD) | Approx. Expected Range3 |
| --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality')+++ |  |  |  |  |
| +++ **INS $**parameter.parameter+++ | +++ **INS $**parameter.units+++ | +++ **INS $**parameter.accuracy+++ | +++ **INS $**parameter.precision+++ | +++ **INS $**parameter.expectedRange+++ |
| +++END-FOR parameter +++ |  |  |  |  |

1“General” accuracy objectives are estimates assuming a true value is known and could be tested; all analytical accuracy objectives (i.e., for samples) include non-detectable concentration in ambient field blanks.

2 For analytical samples, the objective for overall precision is typically based on the relative percent difference (RPD) of co-located, simultaneous duplicates

3 Ranges may vary from proposed. Consult your laboratory and scientific advisory committee and insert the appropriate range for your specific study.

The QA Manager will determine how the resulting dataset compares with the monitoring program’s data quality objectives. This review will include, for each parameter, calculation of the following:

* Completeness goals: overall percent of samples passing QC tests vs. number proposed in Section A7
* Percent of samples exceeding accuracy and precision limits
* Average departure from accuracy and precision targets

After reviewing these calculations and taking into consideration such factors as clusters of unacceptable data (e.g., whether certain parameters, sites, dates, volunteer teams, etc., produced poor results), the Project Manager and/or QA Manager will evaluate the overall attainment of data quality objectives and determine what limitations to place on the use of the data, or if a revision of the data quality objectives is allowable.

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' ,'','') === true+++

## A7 Freshwater/Benthic Data Quality Objectives

Requirements for ensuring that the data are usable for their intended purpose (that is, are of suitable quality) include accuracy, precision, representativeness, comparability, and completeness. When these requirements are met, the final data product is technically defensible. Data elements for this project are discussed in terms of the appropriate characteristics, defined as:

|  |  |
| --- | --- |
| **Accuracy:** | The extent of agreement between a measured value and the true value of interest. |
| **Precision:** | The extent of mutual agreement among independent, similar, or related measurements. |
| **Representativeness:** | The extent to which measurements represent true systems. |
| **Comparability:** | The extent to which data from one study can be compared directly to similar studies. |
| **Completeness:** | The measure of the amount of data acquired versus the amount of data required to fulfill the statistical criteria for the intended use of the data. |

Sections A7.1 and A7.2 describe data quality objectives. Section B5 presents details on how these criteria are met for each component of the program’s monitoring tasks.

### A7.1 Sample Collection Data Quality Objectives

#### Data Quality Objectives by Assessment Type

+++IF determine('Freshwater Benthic', 'Freshwater' ,'Macroinvertebrates', '') === true +++

##### Macrofaunal Sampling Data Quality Objectives

The data quality objectives for the benthic infauna are that (1) transects will be determined appropriately for the stream size and morphology and (2) at least 100 organisms will be collected per sample site.

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' ,'Benthic algal biomass', '') === true +++

##### Benthic Algal Biomass Sampling Data Quality Objectives

Data quality objectives for assessment of benthic algal biomass and algal community are that (1) transects will be determined appropriately for the stream size and morphology; (2) at least 35 observations will be made with each sample site; (3) at least 75% of taxa will be identified; and (4) the identifications correspond to those used throughout the monitoring program, through use of appropriate technical taxonomic literature or other references (e.g., identification keys, voucher specimens).

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' , 'Stream characteristics', '') === true +++

##### Stream Characteristics Assessment Data Quality Objectives

Data quality objectives for physical habitat assessment are that at least 90% of sampling sites will be assessed, and precision in measurements/observations and map-based measurement will be +10%.

+++END-IF+++

#### Accuracy

Each data entry will be checked to the original Field Data Form and random quality control checks are made on subsequent data that have been analyzed or manipulated. The following quality control/quality assurance measures will be taken in the field to ensure accuracy:

+++IF determine('Freshwater Benthic', 'Freshwater' ,

'Macroinvertebrates', '') === true || determine('Freshwater Benthic', 'Freshwater' , 'Benthic algal biomass', '') === true +++

* For macroinfaunal and algal bioassessments, (1) repeat (and/or parallel) field collections and analyses will be performed by separate field crews; (2) occasional alternating and mixing of field personnel will be undertaken to maintain objectivity (minimize individual bias).

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' , 'Stream characteristics', '') === true +++

* For physical habitat assessment, final conclusions are potentially subject to variability among investigators. Ensuring that each investigator is appropriately trained in the evaluation technique, and conducting periodic crosschecks among investigators to promote consistency, will ensure conclusions reached by different investigators are consistent. The crosschecks may consist of comparing rank order of the evaluated sites. That is, rather than comparing the score for each parameter, comparing the total score for each habitat assessment, which yields the rank order of sites (their placement in the assessment from good to bad) for comparison.

+++END-IF+++

#### Representativeness

Representativeness refers to the extent to which measurements represent the true environmental condition*.* Representativeness is affected by the selection of the target surface water bodies, the location of sampling sites within that body, the time period when samples are collected, and the time period when samples are analyzed. The sampling protocols defined for each indicator attempt to address representativeness within the constraints of the sampling design and sampling period.

##### Representativeness—Sampling Design

Benthic monitoring is conducted only in wadeable streams/wadeable reaches of riverine systems. Composite samples from habitat types within the selected reach are assumed representative of that reach. Both active (kick-net) and passive (rock basket) macroinvertebrate and benthic algal biomass sampling increase representation of taxa in the reach.

Due to the way stations are chosen, they are not necessarily representative of typical conditions along an entire stream or wetland complex. A probability‐based sampling design should provide estimates of condition of surface water resource populations that are representative of the region. To minimize effects of habitat heterogeneity, field methods contain protocols for targeting specific habitats to ensure that samples represent standardized conditions.

##### Representativeness—Sampling Period

This sampling protocol does not attempt to collect data that are representative of conditions year-round. Rather, data collection will occur primarily during the summer months (generally June through September; see schedules in Section A6). For stream macroinvertebrates, sampling is conducted July through September when macroinvertebrate communities are at their most active and diverse, and when environmental conditions are at their most stressful, i.e., when low flow and high temperatures combine to stress aquatic life resources. Algal assessments, conducted in June and July, target peak algal growth and minimize confounding effects of spring runoff.

#### Completeness

One hundred percent of samples will be collected and analyzed.

#### Comparability

Standardized sampling and analytical methods, units of reporting, and site selection procedures will be used to ensure comparability of data.

Standardized sampling procedures will be used as described in Section B2. Standardized training, sampling procedures, sampling equipment, and analytical methodologies will be carried out by all sampling crews and laboratories. If some incomparability between sampling crews comes to light, the data collected by those crews will be evaluated and possibly rejected.

Comparability of data within and among indicators is also facilitated by the implementation of standardized quality assurance and quality control techniques and standardized performance and acceptance criteria. For all measurements, reporting units and format are specified, incorporated into standardized data recording forms, and documented in the information management system.

#### Holding Times

Samples that may preserve poorly (e.g., samples collected with a large amount of accompanying sediment or organic material) will be drained of the original ethyl alcohol added in the field and replenished with fresh 95% ethyl alcohol until processed. Sample maintenance will help to prevent sample decomposition or deterioration, as well as keep offensive odors to a minimum.

#### Precision

Appropriate methodology and adequate training and instruction of personnel in methods application will ensure precision.

+++IF determine('Freshwater Benthic', 'Freshwater' ,

'Macroinvertebrates', '') === true+++

### A7.2 Sample Analysis Data Quality Objectives

#### Macrofaunal Analysis Data Quality Objectives

The data quality objectives for the analysis of benthic infauna are that (1) all samples be processed; (2) at least 95% of all animals will be extracted for identification and enumeration; (3) all infauna will be counted accurately (in the field, and confirmed by the taxonomist); (4) the taxonomic identifications be accurate to genus level, and a random 10% of the identified samples are confirmed correct by an independent taxonomist; and (5) the identifications correspond to those used throughout the monitoring program, through use of appropriate technical taxonomic literature or other references (e.g., identification keys, voucher specimens).

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,'', '') === true+++

## A7 Marine/Benthic Data Quality Objectives

Requirements for ensuring that the data are usable for their intended purpose (that is, are of suitable quality) include accuracy, precision, representativeness, comparability, and completeness. When these requirements are met, the final data product is technically defensible. Data elements for this project are discussed in terms of the appropriate characteristics, defined as:

|  |  |
| --- | --- |
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| **Precision:** | The extent of mutual agreement among independent, similar, or related measurements. |
| **Representativeness:** | The extent to which measurements represent true systems. |
| **Comparability:** | The extent to which data from one study can be compared directly to similar studies. |
| **Completeness:** | The measure of the amount of data acquired versus the amount of data required to fulfill the statistical criteria for the intended use of the data. |

Data quality objectives are given below. Details of how these criteria are met for each component of the benthic monitoring tasks are presented in Section B5.

+++IF determine('Saltwater Benthic', 'Saltwater' , 'Sample penetration depth', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Grab sample volume', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Sediment texture', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Grain size', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Total organic carbon (TOC)', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Infauna', '') === true +++

### A7.1 Benthic Grab Sampling—Data Quality Objectives (DQOs)

The data quality objectives for collection of sediment grab samples are that (1) samples be collected within 30 meters of the target location; (2) all required samples be collected; (3) samples be of sufficient quantity to be representative of the station; (4) samples be undisturbed; and (5) samples be uncontaminated.

The determination of sufficient quantity is made by measuring the depth of penetration of the grab. The 0.04 m2 Young-modified Van Veen grab sampler used for sediment and biological samples must contain sediment to a depth of at least 7 cm (out of a possible 10 cm) at the center. The sediment within the sampler must have a relatively level surface and not spill out from the top of the grab (Figure A7.1). Procedures for collecting undisturbed and uncontaminated samples are described in Section B2.

The data quality objectives for the handling of sediment samples to be used for sedimentary analysis are that (1) samples remain uncontaminated, (2) samples be well homogenized, and (3) samples be subsampled and preserved following methods detailed in Section B2.

The data quality objectives for handling benthic infaunal samples are that (1) samples be handled gently during the sieving process, (2) samples be fixed in alcohol as quickly as possible to prevent deterioration of the fauna, and (3) sample jars be labeled accurately. Procedures for sample handling are detailed in Section B3.

All sediment samples analyzed for grain size and total organic carbon (TOC) will follow the NCCA Laboratory Analysis Requirements.[[1]](#footnote-2) Samples for grain size analysis will be refrigerated at 4 °C and analyzed with any method that characterizes the sediments and meets QA/QC requirements. TOC samples will be frozen at a maximum of -20 °C and analyzed using the Lloyd Kahn Method.



Figure A7.1. Illustration of acceptable and unacceptable grabs for benthic community analysis using a 0.04 m2 Van Veen grab sampler.[[2]](#footnote-3)

### 

### A7.2 Infaunal Analysis Data Quality Objectives

The data quality objectives for the analysis of benthic infauna are that (1) all samples be processed; (2) all animals be removed for identification and enumeration via the sorting protocol; and (3) all infauna be counted accurately. The laboratory QAPP (attached) includes the following data quality objectives: (1) the taxonomic identifications in the laboratory be accurate (correct) and (2) the identifications correspond to those used throughout the monitoring program. At least 95% of all animals must be removed from a sample to pass the quality control (QC) evaluation as discussed in Section B5.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' , 'Sample penetration depth', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Grab sample volume', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Sediment texture', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Infauna', '') === true +++

### A7.1 Benthic Grab Sampling—Data Quality Objectives

The data quality objectives for collection of sediment grab samples are that (1) samples be collected within 30 m of the target location; (2) all required samples be collected; (3) samples be of sufficient quantity to be representative of the station; (4) samples be undisturbed; and (5) samples be uncontaminated.

The determination of sufficient quantity is made by measuring the depth of penetration of the grab. The 0.04-m2 Ted Young-modified van Veen grab sampler used for sediment and biological samples must contain sediment to a depth of at least 7 cm (out of a possible 10 cm) at the center. The sediment within the sampler has a relatively level surface and is not spilling out from the top of the grab (Figure 1). Procedures for collecting undisturbed and uncontaminated samples are described in Section B2.

The quality objectives for the handling of benthic infaunal samples are that (1) samples be handled gently during the sieving process, (2) samples be fixed in alcohol as quickly as possible to prevent deterioration of the fauna, and (3) sample jars be labeled accurately. Procedures for sample handling are detailed in Section B3.



Figure A7.1. Illustration of acceptable and unacceptable grabs for benthic community analysis using a 0.04 m2 Van Veen grab sampler.[[3]](#footnote-4)

### A7.2 Infaunal Analysis Data Quality Objectives

The data quality objectives for the analysis of benthic infauna are that (1) all samples be processed, (2) all animals be removed for identification and enumeration, (3) all infaunal animals be counted accurately, (4) the taxonomic identifications in the laboratory be accurate (correct), and (5) the identifications correspond to those used throughout the monitoring program. At least 95% of all animals must be removed from a sample to pass the quality control (QC) evaluation as discussed in Section B5.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,'Grain size', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Total organic carbon (TOC)', '') === false +++

### A7.1 Benthic Grab Sampling—Data Quality Objectives (DQOs)

The quality objectives for collection of sediment grab samples are that (1) samples be collected within 30 m of the target location, (2) all required samples be collected, (3) samples be of sufficient quantity to be representative of the station, (4) samples be undisturbed, and (5) samples be uncontaminated.

The determination of sufficient quantity is made by measuring the depth of penetration of the grab. The 0.04 m2 Ted Young-modified van Veen grab sampler used for sediment and biological samples must contain sediment to a depth of at least 7 cm (out of a possible 10 cm) at the center. The sediment within the sampler has a relatively level surface and is not spilling out from the top of the grab (Figure A7.1). Procedures for collecting undisturbed and uncontaminated samples are described in Section B2.

Figure A7.1. Illustration of acceptable and unacceptable grabs for benthic community analysis using a 0.04 m2 Van Veen grab sampler.[[4]](#footnote-5)

The data quality objectives for the handling of sediment samples to be used for sedimentary analysis are that (1) samples remain uncontaminated, (2) samples be well homogenized, and (3) samples be subsampled and preserved following methods detailed in Section B2.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,'Grain size', '') === true && determine('Saltwater Benthic', 'Saltwater' , 'Total organic carbon (TOC)', '') === true +++

### A7.1 Benthic Grab Sampling—Data Quality Objectives

The data quality objectives for collection of sediment grab samples are that (1) samples be collected within 30 m of the target location, (2) all required samples be collected, (3) samples be of sufficient quantity to be representative of the station, (4) samples be undisturbed, and (5) samples be uncontaminated.

The determination of sufficient quantity is made by measuring the depth of penetration of the grab. The 0.04 m2 Ted Young-modified Van Veen grab sampler used for sediment and biological samples must contain sediment to a depth of at least 7 cm (out of a possible 10 cm) at the center. The sediment within the sampler must have a relatively level surface and not spill out from the top of the grab (Figure A7.1). Procedures for collecting undisturbed and uncontaminated samples are described in Section B2.

Figure A7.1. Illustration of acceptable and unacceptable grabs for benthic community analysis using a 0.04 m2 Van Veen grab sampler.[[5]](#footnote-6)

The data quality objectives for the handling of sediment samples to be used for sedimentary analysis are that (1) samples remain uncontaminated, (2) samples be well homogenized, and (3) samples be subsampled and preserved following methods detailed in Section B2.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,'Grain size', '') === false && determine('Saltwater Benthic', 'Saltwater' , 'Total organic carbon (TOC)', '') === true +++

### A7.1 Benthic Grab Sampling—Data Quality Objectives

The data quality objectives for collection of sediment grab samples are that (1) samples be collected within 30 m of the target location, (2) all required samples be collected, (3) samples be of sufficient quantity to be representative of the station, (4) samples be undisturbed, and (5) samples be uncontaminated.

 The determination of sufficient quantity is made by measuring the depth of penetration of the grab. The 0.04 m2 Ted Young-modified van Veen grab sampler used for sediment and biological samples must contain sediment to a depth of at least 7 cm (out of a possible 10 cm) at the center. The sediment within the sampler must have a relatively level surface and not spill out from the top of the grab (Figure A7.1). Procedures for collecting undisturbed and uncontaminated samples are described in Section B2.

Figure A7.1. Illustration of acceptable and unacceptable grabs for benthic community analysis using a 0.04 m2 Van Veen grab sampler.[[6]](#footnote-7)

The data quality objectives for the handling of sediment samples to be used for sedimentary analysis are that (1) samples remain uncontaminated, (2) samples be well homogenized, and (3) samples be subsampled and preserved following methods detailed in Section B2.

+++END-IF+++

+++END-IF+++

+++IF determine('Saltwater Water Quality','Saltwater' ,'','') === true+++

## A7 Marine/Water Quality Data Quality Objectives

Requirements for ensuring that the data are usable for their intended purpose (that is, are of suitable quality) include accuracy, precision, representativeness, comparability, and completeness. When these requirements are met, the final data product is technically defensible. Data elements for this project are discussed in terms of the appropriate characteristics, defined as:

|  |  |
| --- | --- |
| **Accuracy:** | The extent of agreement between a measured value and the true value of interest. |
| **Precision:** | The extent of mutual agreement among independent, similar, or related measurements. |
| **Representativeness:** | The extent to which measurements represent true systems. |
| **Comparability:** | The extent to which data from one study can be compared directly to similar studies. |
| **Completeness:** | The measure of the amount of data acquired versus the amount of data required to fulfill the statistical criteria for the intended use of the data. |

A data quality objectives table accompanies this text. Details of how these criteria are met for each component of the benthic monitoring tasks are presented in Section B5.

Table A7.3.  Data Quality Objectives for Marine Water Quality

| Parameter | Units | Accuracy1 (+/-) | Overall Precision2 (RPD) | Approx.  Expected Range3 |
| --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality')+++ |  |  |  |  |
| +++ **INS $**parameter.parameter+++ | +++ **INS $**parameter.units+++ | +++ **INS $**parameter.accuracy+++ | +++ **INS $**parameter.precision+++ | +++ **INS $**parameter.expectedRange+++ |
| +++END-FOR parameter +++ |  |  |  |  |

1“General” accuracy objectives are estimates assuming a true value is known and could be tested; all analytical accuracy objectives (i.e. for samples) include non-detectable concentration in ambient field blanks.

2 For analytical samples, the objective for overall precision is typically based on the relative percent difference (RPD) of co-located, simultaneous duplicates

3 Ranges may vary from proposed. Consult your laboratory and scientific advisory committee and insert the appropriate range for your specific study.

MDL = Method Detection Limit (Lab)

RDL = Reporting Detection Limit (Lab)

+++END-IF+++

## A8 Training Requirements

Training on all aspects of project data collection and management will be provided to project participants and will be documented—including trainee signatures, trainer signatures, dates of training, and subject matter—in a training log.

All members of the project team will be required to attend training/workshops appropriate to the type of monitoring they will conduct. The Project Manager shall ensure that volunteers receive appropriate training by organizing and conducting workshops (securing the services of expert trainers as needed) and/or arranging for volunteers to be trained at workshops held by other qualified personnel or organizations.

The project manager enters training data into the project database and records the following information: subject matter (i.e., what type of monitoring and procedures are covered), training course title, type of training materials, date and agenda, name and qualification of trainers, and names of participants trained. Volunteers shall be trained in monitoring protocols and be able to document pertinent environmental data for the evaluation site. Project-specific training activities are included as an attached form.

All training activities will be documented by training forms signed by the trainees (form attached), with documentation in a final report.

## A9 Documentation and Records

### A9.1 Documentation

Initially, all data will be recorded onto established data forms. All data collection notes will be made in permanent ink, initialed, and dated, and no erasures or obliterations will be made. Completed data forms or other types of hand-entered data will be signed and dated by the individual entering the data. Data will be recorded electronically onto computer storage media. Direct-entry and electronic data entries will indicate the person collecting or entering the data. Secondary data used will be documented in the Secondary Data Form, attached.

Table A9.1. Record Handling Procedures

|  |  |
| --- | --- |
| Activity | Details |
| Document and record-keeping | Where will records (forms) be stored? How long will they be retained? |
| Data generation | What records will be produced – field, lab, and/or assessment? |
| Data quality package | Final project QAPP including attachments and corrective actions, documentation of any substantive changes made to the AquaQAPP-generated QAPP, formatted data, data evaluation summary |
| Data reporting format | Digital file type? |
| Data storage | Where will the digital data be stored? EPA’s Water Quality Exchange (WQX) database is a suitable response here. |

### A9.2 Field Records

Field Data Forms will provide the primary means of recording the data collection activities performed during the sampling surveys. As such, entries will be described in as much detail as possible so that events occurring the survey can readily be reconstructed after the fact. At the beginning of each survey, the date, start time, weather, and names of all sampling team members present will be entered, along with information about the samples, on a Field Data Form

Table A9.2. Project-Specific Datasheets, Labels, and Forms for All QAPPs

| Document Type | Form Name | Description | Relevant to which QAPP |
| --- | --- | --- | --- |
| Field data sheets | Field Data Form | Completed on site at the time of sampling. Includes field measurement (e.g. YSI) and sample collection info, site location and ID, crew names, weather conditions, etc. | All |
| Site verification form | Site Assessment Form | Completed as part of site assessment verification visit | All |
| Equipment Custody Form | Equipment Custody Form | Lists all equipment provided to volunteers. Completed by volunteers upon receipt of sampling kit. Serves as checklist of equipment provided for tracking. | All |
| Laboratory Data Sheets | Laboratory Data Form | These forms will include lab SOP number, data analysis, QA/QC and results. | All |
| Chain-of-Custody Forms | Chain-of-Custody Form | These will accompany samples from collection sites to labs. Will include sample tracking to labs. | All |
| Sample labels | Sample Label | These will be placed on all sample containers and will include the site ID, date, time, parameter to be analyzed, and monitor’s initials. | All |
| Training and Evaluation Form | Training and Evaluation Form | These will include information on volunteer training | All |
| Instrument Log | Instrument Calibration Log | This will include information on maintenance, calibration and testing on equipment | All |
| Sample Checklist | Sample Log | This will maintain a list of samples collected in the field. | All |

+++IF determine('Saltwater Benthic','Saltwater','','') === true && determine('Freshwater Benthic','Freshwater','','') === true+++

Table A9.2. Project-Specific Datasheets, Labels, and Forms for All Benthic QAPPs

| Document Type | Form Name | Description | Relevant to which QAPP |
| --- | --- | --- | --- |
| Photo log | Photo Log Form | Completed on site at the time of sampling | Freshwater benthic, marine benthic |

+++END-IF+++

+++IF determine('Freshwater Benthic','Freshwater','','') === true+++

Table A9.2. Project-Specific Datasheets, Labels, and Forms for Freshwater Benthic QAPPs

| Document Type | Form Name | Description | Relevant to which QAPP |
| --- | --- | --- | --- |
| Flow velocity estimation | Flow Velocity Form | Completed on site at the time of sampling | Freshwater benthic |
| Algal biomass field sheet | Algal Biomass Field Form | Completed on site at the time of sampling | Freshwater benthic |
| Algal biomass reference sheet | Algal Biomass Reference Sheet | Preparatory material before field visit | Freshwater benthic |
| Kick sample collection filed sheet | Kick Sample Field Data Form | Completed on site at the time of sampling | Freshwater benthic |
| Rock basket field sheet | Rock Basket Field Data Form | Completed on site at the time of sampling | Freshwater benthic |
| Invertebrate sorting sheet | Invertebrate Sorting Form |  | Freshwater benthic |
| Habitat assessment field sheet | Habitat Assessment Field Form | Completed on site at the time of sampling | Freshwater benthic |

+++END-IF+++

Information specific to sample collection will include:

* Station name and/or ID number
* Replicate number
* Time and date of sample collection
* Sample description (color, texture, etc.)
* Samplers’ initials
* Requested analyses
* Location (the geographic location where a sample is collected)

Supplementary data for every station sampled during the field surveys may be recorded in the comments section of the Field Data Form. Additional data may include notes on sampling difficulties, currents, presence/absence of anemones, and numbers and sizes of jars used for each sample.

### +++IF determine('Saltwater Benthic','Saltwater','Grain size','Sediment grab samples') === true && determine('Saltwater Benthic','Saltwater','Total organic carbon (TOC)','Sediment grab samples') === true && determine('Saltwater Benthic','Saltwater','Infauna','Sediment grab samples') === true+++A9*.*3 Data Analyses—Marine Benthic Grab - if all marine benthic parameters selected

The sediment data will be analyzed using a variety of statistical and graphical methods. Various univariate and multivariate analyses may be employed using a standard statistical/graphics package (e.g., R, SAS, PRIMER). These tests may include analysis of variance (ANOVA), correlation analyses, or regression analyses. Additional evaluations may assess temporal and spatial trends in sediment data as compared to benthic faunal distributions.

Prior to analysis of the infaunal data, some modifications to the dataset will be made. For example, some taxa (e.g., incidental pelagic faunal, encrusting, or non-benthic taxa) may be eliminated from all calculations. Other taxa may be included in calculations of abundance but not diversity; such taxa are usually those infaunal organisms that cannot be identified to species level. Only those individuals identified to species level will be included in all remaining calculations (e.g., number of species, diversity, evenness, multivariate analyses).

Two will be calculated: (1) species richness indices and (2) indices based on the proportional abundances of species—i.e., Shannon-Weiner index (H’), Pielou evenness index (J’), Margalef’s index, and/or Total Taxonomic Distinctness).

Changes in infaunal community structure between assessments may be evaluated by comparing community structure differences between stations through time, and gauging changes in community structure if comparable data are available.

+++END-IF+++

+++IF determine('Saltwater Benthic','Saltwater','Infauna','Sediment grab samples') === true+++

### A9.4 Infaunal Data Analyses—Marine Benthic Grab - If only the marine benthic infaunal group selected

Prior to analysis of the infaunal data, some modifications to the dataset will be made. For example, some taxa (e.g., incidental pelagic faunal, encrusting, or non-benthic taxa) may be eliminated from all calculations. Other taxa may be included in calculations of abundance but not diversity; such taxa are usually those infaunal organisms that cannot be identified to species level. Only those individuals identified to species level will be included in all remaining calculations (e.g., number of species, diversity, evenness, multivariate analyses).

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater' ,'','') === true+++

# Section B. Fresh Water/Water Quality Data Generation and Acquisition

## B1 Sampling Design

+++IF determineConcern('Eutrophication (Nutrients)') === true || determineConcern('Illicit Connections') === true || determineConcern('Stormwater') === true +++

### B1.1 Sampling Site Selection

For point source assessments, the sampling site selection will include stations upstream and downstream of the source, as well as reference stations. The site selected will be in an area where reasonable opportunity for mixing of the effluent has occurred. Where a mixing zone has been defined in a license, sampling will be conducted immediately downstream of it. In cases where the effluent plume channels down one bank for great distances (>1 km), or where localized effluent impact is expected to be severe for a distance beyond the zone of initial dilution, sampling at a site upstream of the source, one or more in the plume, and at least two farther downstream will take place. One downstream site will be located at the point of presumed bank to bank mixing and subsequent sites will be located to assess the extent of impact downstream. The area of initial dilution of an effluent will be determined by visual observation of the plume pattern; by observations of biotic effects attributable to the plume, if evident (benthic algal biomass growth, die-off patterns); and by transects of conductivity (specific conductance) measurements from the outfall, in a downstream direction.

Sampling sites will be selected to ensure that the physical characteristics among sampling sites are similar. For surveys conducted to determine use designations, sampling locations will be representative of the stream reach. Reference conditions will include minimally impaired sites in the same ecoregion, size class, and stream type (width, depth, gradient).

+++END-IF+++

+++IF determineConcern('General Environmental Health: Physical/Chemical Water Quality') === true || determineConcern('Recreation (Swimming and/or Boating)') === true || determineConcern('Harmful Algal Blooms (HABs) (Algal toxins)') === true || determineConcern('General Environmental Health: Benthic') === true || determineConcern('Acidification') === true +++

### B1.1 Sample Site Selection

For water quality condition assessment, routine sampling activities will consist of collecting in-stream samples. Routine sampling will be representative of overall water quality and are relatively unchanging over time to allow comparison to past and future investigations. Sites will be generally selected at the downstream ends and/or key segmentation points of major tributaries and at or near locations where there is a longstanding data record.

To meaningfully evaluate water quality condition, sampling locations will be selected to ensure generally comparable physical habitat. Sample locations will be scouted out ahead of time to identify appropriate reaches and accessibility for sampling. Station siting of both study sites and reference sites will take place during June. Reconnaissance activities prior to June are not desirable, as instream conditions—most notably flow regimes—during this time are often dramatically different than during the sampling index period. To lay out the sampling reach and sites, the route to the site will be explored to ensure it is free of obstacles that would prohibit sampling and data collection activities, then the sample reach characteristics will be assessed. Field conditions (e.g., instream and riparian habitat characteristics, surrounding land use, observations of nonpoint source pollution or other pertinent information) during the time of reconnaissance will be noted and recorded in a field notebook.

+++END-IF+++

### B1.2 Location

See Section A6. A map with sampling locations labeled is included as an attachment. The map includes a legend, scale, and compass direction.

[insert summary table – site id, name, parameter(s)]

### B1.3 Sample Collection Methods

Sample types include grab samples and direct measurements using electronic instruments in the field. Water quality parameters that are measured/observed in situ as well as indicators to be analyzed in the laboratory are listed in the table below.

Routine sampling events are generally scheduled every four weeks, starting in May through October. This will meet the new Surface Water Quality Standards for bacteria which stipulate that at least five samples be taken within a six-month period to make a use determination. Field duplicates are routinely collected for quality control purposes. The details of the sampling design are described in the table below.

Table B1.1. River and Stream Field Sampling Summary

| Parameter - Method | Number of sample locations (At least one each for selected reach or tributary) | Rationale for number of samples | Site location rationale | Frequency | Number/type of QC samples including field duplicates (10%) and blanks (10%) |
| --- | --- | --- | --- | --- | --- |
| +++FOR parameter IN sampleDesign.filter((param) => param.monitoringCategory === 'Freshwater Water Quality') +++ |  |  |  |  |  |
| +++ **INS $**parameter.sampleParameter+++ | +++ **INS $**parameter.numSampleLocations+++ | |  | | --- | | +++ **INS $**parameter.sampleNumRationale+++ | | +++ **INS $**parameter.locationRationale+++ | +++ **INS $**parameter.frequency+++ | +++ **INS $**parameter.numQcSamples+++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

1Dropdown:

* Random or probabilistic
* Accessibility considerations
* Proximity to potential pollutant source
* Replication of previous sampling efforts (*e.g.*, by DEP or EPA)
* Other (please specify): \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

2Dropdown:

* Spatial coverage of waterbody
* Feature of interest
* Regulatory requirement
* Proximity to impact or suspected pollution source
* Capacity (funding or staffing) Replication of previous sampling efforts (*e.g.*, by DEP or EPA)
* Other (please specify): \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

## B2 Sampling Methods: Sample Collection and Storage

Table B2.1. Equipment Preparation, Sample Processing, and Storage Requirements

|  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- |
| Parameter - Method | Sample collection method | Sample Container | Sample Volume | Sample Preservation | Maximum Holding Time |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality')+++ |  |  |  |  |  |
| +++ **INS $**parameter.label+++ | +++ **INS $**parameter.sampleCollectionMethod+++ | +++ **INS $**parameter.sampleContainer+++ | +++ **INS $**parameter.sampleVolume+++ | +++ **INS $**parameter.samplePreservation+++ | +++ **INS $**parameter.maxHoldingTime +++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

\*Pre-cleaned – acid washed with 10% HCL

\*\*in situ: single and/or multiple probe

### B2.1 Water Quality Monitoring

#### Equipment/Instrument Calibration

Prior to field use, the multi-parameter or individual sensors will be calibrated in accordance with the manufacturer’s instruction manual. If no instructions specific to the instrument are available, the following general calibration methods will be followed.

##### General Calibration Methods: For Multi-Parameter Unit or Individual Units

Supply list for taking measurements and calibrating the multi-parameter unit or sonde:

* Multi-parameter water quality unit (with cable and handheld data logger)
* Extra batteries
* De-ionized and tap water in squirt bottles
* Calibration cups and standards
* Dry wipes or paper towels
* Holosteric barometer or elevation chart to use for calibration
* National Institute of Standards and Technology–certified thermometer
* Large bucket of river water
* Calibration records form

Calibration standards:

* pH 7.00 standard buffer solution
* pH 4.00 standard buffer solution
* pH 10.00 standard buffer solution
* 1 mS/cm (1,000 μS/cm) conductivity standard
* Sodium sulfite solution (0% dissolved oxygen)

The following items will also be needed for recording measurements:

* Field Data Form
* Pencils (for data forms)

##### Equipment Calibration Method: For Multi-Parameter Unit or Individual Units

###### Temperature Sensor

Check the accuracy of the sensor against a calibrated thermometer at least once per sampling season.

###### pH Sensor

*Calibration standards required:* pH 4.00, 7.00, and 10.00 standard buffer solutions.

Calibrate the pH sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions and existing standard operating procedures. Ideally, use a QC solution that is similar in ionic strength to the water samples you will be measuring.

*General calibration method:*

1. Use small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with dry wipes or paper towel.
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration.

Record the calibration solution lot number and expiration date on the instrument calibration log.

###### Dissolved Oxygen Sensor

*Calibration standard required:* Sodium sulfite solution (0% dissolved oxygen).

Calibrate the dissolved oxygen unit prior to each sampling event. It is recommended that the sensor probe be calibrated in the field against an atmospheric standard (e.g., ambient air saturated with water). Follow your manufacturer’s guidelines for calibration of the dissolved oxygen probe.

*General calibration method:*

1. Use small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with dry wipes or paper towel. (Never touch the membrane of the dissolved oxygen sensor.)
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration.

Record the calibration solution lot number and expiration date on the instrument calibration log.

###### Conductivity Sensor

*Calibration standard required:* 1 mS/cm (1,000 μS/cm) conductivity standard.

Calibrate the conductivity sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions. Ideally, use a QC solution that incorporates the entire expected conductivity range.

*General calibration method:*

1. Use small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with dry wipes or paper towel.
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration.

Record the calibration solution lot number and expiration date on the instrument calibration log.

###### Turbidity Sensor

*Calibration standard required:* 126 NTU turbidity standard

Calibrate the turbidity sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions.

*General calibration method:*

1. Use small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with dry wipes or paper towel.
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration.

Record the calibration solution lot number and expiration date on the instrument calibration log.

###### Using the Data Logger

Calibration can also be done using the data logger. Follow the instruction manual to calibrate the instrument. Collect river water in a large bucket and let it stand at room temperature for several hours to let the temperature stabilize. Attach the data logger to the sonde with field cable and follow the instruction manual.

#### Multi-Parameter Unit Deployment and Grab Sample Collection

Measurement of temperature, conductivity, dissolved oxygen, and pH will be taken at surface. A hydrographic profile at each site will be obtained at water depth greater than or equal to 2 m. These parameters are measured to detect extremes in conditions that might indicate impairment and depth at location. In situ measurements will be made using a calibrated water quality multi‐parameter unit sonde at each station. Measurements will be collected as the sensor or sonde is lowered, at prescribed intervals (usually 0.5 m to 1.0 m depending on depth) down to 0.5 m from the bottom.

##### Pre-sampling Site Assessment

1. Estimate the total water depth at the sampling site by lowering the depth sounding line (marked in ft) to the bottom of the river and counting the number of taped 1 ft marks on the cable. If possible, collect sensor measurements during the downcast from near surface (approximately 0.5–1.5 m) to near bottom (about 0.5 m off the bottom) or along a hydrographic profile.

Visually scan station for best wade-in area that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If there is no water in the stream, record as “no flow.”

Note: If water depth (ft) × velocity (fps) is over 10, the area is not considered wadable for sampling. Readings can be taken at the bottle sampling site if the river is shallow or from a bridge near the sampling site if the water is too deep to safely get into the main flow of the river. Where wading is not possible to reach the sampling site, sample taken from the bank or from a bridge will be taken in the flow and representative of the larger area. If a representative location cannot be found, no sample will be collected.

##### Method: In Situ Sampling Procedures

1. Take the measurements mid-channel at the sampling site. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
2. Wade in and deploy the multi-parameter unit to let it equilibrate sitting on the river bottom in a location upstream of the wader’s position. If the current is swift, allow the sonde to lie along the bottom to stay submerged.
3. Measure the site depth accurately before taking the measurements. If the depth at the index site is less than 1 m, take the measurements at mid-depth. If conductivity readings are zero, check that the probes are covered with water.

Wait for the readings to stabilize (sometimes as much as 2–3 minutes) and record reading position, total depth and reading depths, time, and readings on the Field Data Form.

+++IF determine('Freshwater Water Quality', 'Freshwater','Total phosphorus', '') === true || determine('Freshwater Water Quality', 'Freshwater',' Total nitrogen', '') === true || determine('Freshwater Water Quality', 'Freshwater',' Ammonia-N', '') === true || determine('Freshwater Water Quality', 'Freshwater','Nitrate-Nitrite-N', '') === true || determine('Freshwater Water Quality', 'Freshwater','Orthophosphate', '') === true+++

### B2.2 Nutrients

Supply list for collecting samples:

* Nitrile gloves
* 4 L sample container
* 2 L amber Nalgene bottle
* Cooler with ice
* Dry ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

***Pre-sampling Site Assessment***

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” As a rule of thumb (USGS), do not wade in to sample if water depth (ft) × velocity (fps) is over 10. Where wading into the flow is not possible for the chosen site and no alternative is available, ensure that a sample taken from the bank is in the flow and representative of the larger area. If a representative location cannot be found, do not take sample.

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still).
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Sample Storage and Handling

1. Place the bottles in a cooler (on ice or water) and shut the lid.
2. Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
3. At the lab, store samples in a refrigerator at 4°C and process within 24 hours. If not analyzed within this time, the samples should be filtered and the filters frozen for future analysis (within 21 days of their collection).

If samples cannot be delivered to the laboratory within 6–8 hours following collection, acid preservation may be required. See “Method: Processing Nutrient Samples.”

#### Method: Processing Nutrient Samples

1. Before acidifying samples, put on disposable gloves and safety glasses.
2. Remove acid bottle from the ziploc bag, and carefully draw approximately 1 mL of 9N sulfuric acid per 250 mL (e.g., 2 mL for 500 mL sample) and dispense into sample to achieve sample pH < 2.

Cap sample and mix thoroughly. Carefully recap acid and discard used pipette into separate acid-refuse bag. Place samples back on ice.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Chlorophyll-a', '') === true+++

### B2.3 Chlorophyll *a*

Supply list for collecting samples:

* Nitrile gloves
* 250 L amber Nalgene bottle
* Cooler with ice
* Dry ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” As a rule of thumb (USGS), do not wade in to

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Sample Storage and Handling

* No chemical preservation in the field is needed.
* Place the bottles in a cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store samples in a refrigerator at 4°C and process them within 24 hours of the collection time.
* If not analyzed within this time, the samples will be filtered and the filters frozen for future analysis (within 21 days of their collection). See “Method: Filtering Chlorophyll *a* Samples.”

#### Method: Filtering Chlorophyll a Samples

1. In low light conditions, set up the filter apparatus with vacuum flask, filter holder, glass fiber filter, and filter funnel.
2. Using a clean graduated cylinder, measure a precise volume and record the amount on the Field Data Form.
3. Pour the measured sample into the clean filter funnel and filter with a vacuum pump (electric pump or by hand). Filter at least 500 mL of the sample. A good guide is a visible quantity of green or greenish brown on the filter. Note: If you don’t see more than a tinge, filter more sample. Filtering may significantly slow in the later stages as the filter plugs up with material.
4. Record the volume filtered to the nearest mL.
5. Remove the filter funnel, and carefully remove the filter from the filter holder using forceps. Fold the filter in half (green side in), and place in an air drying box and cover.
6. Rinse all equipment (cylinder, filtering apparatus, and forceps) with distilled water before processing additional samples.

When all samples have been filtered, plug in the drying box. Air dry the sample filters for at least 45 minutes or until they are dry. Remove filters with forceps and place in aluminum foil. Label the aluminum foil; prepare for delivery to the laboratory or freeze.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Chlorides','') === true+++

### B2.4 Chlorides

Supply list for collecting samples:

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Do not wade in to

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Sample Storage and Handling

* Place the bottles in a cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store in a refrigerator at 4°C and process within 28 days.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Turbidity', '') === true+++

### B2.5 Turbidity

Supply list for collecting samples:

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Do not wade

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Sample Storage and Handling

* Place the bottles in a cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store samples in a refrigerator at 4°C and process within 48 hours.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Total suspended solids', '') === true+++

### B2.6 Total Suspended Solids

Supply list for collecting samples:

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Do not wade in to

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Sample Storage and Handling

* Place the bottles in a cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store samples in a refrigerator at 4°C and process within seven days.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Microcystins', '') === true+++

### B2.7 Algal Toxins—Microcystins

The algal toxin (microcystin) sample is a grab sample taken from the site. The grab sample is collected using the 3 L beaker to fill two 500 mL bottles. A screening test is conducted in the field using dipsticks. If the presence of microcystins is detected, and concentration is higher than acceptable, the sample may be taken to the laboratory for further analysis.

Supply list for collecting samples:

* Nitrile gloves
* 3 L Nalgene beaker
* HDPE bottle (500 mL white, round)
* Plastic electrical tape
* Cooler with ice
* Algal toxin strip test kit for microcystins

For recording measurements:

* Sample collection form
* Sample labels with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Do not wade

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.

Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.

To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).

Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.

To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

#### Field Method: Algal Toxin Strip Test for Microcystin

1. Pour 1–2 mL of the sample from the HDPE bottle into the small bottle provided with the test kit.
2. Using the graduated pipette provided, transfer 1 mL of sample to the lysis vial containing the dried lysis reagent.
3. Cap the bottle and shake for 2 minutes. Let rest for 8 minutes.
4. Using the forceps provided, add 1 reagent paper to the lysis vial. Cap and shake for 2 minutes. Let rest for 8 minutes.
5. Using the pipette provided, add 7 drops of sample to the conical, flip-top tube containing the reagent.
6. Close the conical, fliptop tube and shake it for 30 seconds. Sample will turn purple.
7. Insert test strip into conical, fliptop tube with arrow pointing down (sample pad down). Incubate for 10 minutes.
8. Remove test strip. Lay flat and allow to continue developing for 5 minutes.

Use the strip control and test lines to measure approximate concentration of microcystins observed.

#### Sample Storage and Handling

* Place the 500 mL bottles in a cooler (on ice or water) and shut the lid. If a cooler is not available, place the 500 mL bottles in an opaque garbage bag and immerse them in the stream.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* Upon returning to your base site, freeze sample and keep frozen until shipping. Mark the “Frozen” bubbles on the form to verify samples have been frozen.

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'E. coli', '') === true+++

### B2.8 Fecal Indicator—*E. Coli*

Supply list for collecting samples:

* Nitrile gloves
* Pre-sterilized, 120 mL HDPE sample bottle
* Sodium thiosulfate tablet
* Wet ice
* Cooler

For recording measurements:

* Sample collection forms
* Pencils

#### Method: General Bottle Sampling Method

A bacteria group grab sample is collected in a sterile, 120 mL HDPE container with an attached, secure-lock HDPE cap. Containers should have a sodium thiosulfate tablet included for dechlorination, if ambient water at sampling locations possibly contains residual chlorine. Collect the bacterial sample after all other sampling is completed. Filters must be frozen within 6 hours of collection.

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Do not wade in to sample if water depth (ft) × velocity (fps) is over 10. Where wading into the flow is not possible for the chosen site and no alternative is available, ensure that a sample taken from the bank is in the flow and representative of the larger area. If a representative location cannot be found, do not take sample.

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams Using Grab Samples

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Select a sampling location approximately 1 m from the bank and approximately 0.3 m deep. Approach the sampling location slowly from downstream or downwind. (If the depth does not reach 0.3 m at 1 m from the bank, take the sample and flag it on the field form.)
3. Put on sterile nitrile gloves.
4. Lower the uncapped, inverted 120 mL sample bottle to a depth of 6 in. below the water surface, avoiding surface scum, vegetation, and substrates.
5. Point the mouth of the container away from your body and the boat. Right the bottle and raise it through the water column, allowing bottle to fill completely. If the depth does not reach 0.3 m at 1 m from the bank, take the sample and flag it on the field form.
6. After removing the container from the water, discard a small portion of the sample to allow for proper mixing before filtering (down to the 120 mL mark on the bottle).

Add the sodium thiosulfate tablet, cap, and shake bottle 25 times.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place a new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to the laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Rinse each water sample collection container and lid three times with water. Discard the rinsate downstream.
3. Place the bottle in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
4. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth and then rotate the rod to fill the bottle.
5. Once the bottle is full, remove from the water, discard a small portion of the sample to allow for proper mixing before filtering (down to the 120 mL mark on the bottle).
6. Add the sodium thiosulfate tablet, cap, and shake bottle 25 times.
7. Place on ice to 4°C immediately.
8. To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take a sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper Than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 inches, and then proceed as follows from the upstream side of the drop/bridge:

1. Use sterile, 1,000 mL narrow mouth thiosulfate or non-thiosulfate bottles.
2. Rinse basket three times without bottles. Place required weight in basket as appropriate (using sample bottles filled with sand).
3. Fit the weighted basket sampler with 500 mL narrow-mouth bottles (some filled with sand, others empty, depending on number of samples needed), including sterile ones for bacteria collection.
4. Secure bottles inside basket with mini-bungees. Break sterile seal and uncap bottles (except any filled with sand). Do not use container if sterile seal is not secure.
5. Do not rinse insides of bottles. Lower slowly to water surface and gently plunge into water to approximately 6 in. below water surface. Allow bottles to fill.
6. Observe the sampler closely to ensure that it does not touch bottom sediments.
7. When bubbling has almost stopped, pull up basket slowly. While pulling the basket up, note if any debris from the bridge or the tow line/rope is falling into the sample bottles.
8. Transfer to the smaller, thiosulfate-containing, 120 mL HDPE bottle. (Cap and invert to mix contents of 1,000 mL container; uncap and pour into the sterile 120 mL bottle. Discard a small portion of the sample to allow for proper mixing down to the 120 mL mark on the bottle). Add the sodium thiosulfate tablet, cap, and shake bottle 25 times.
9. Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to the laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than 18 in. approximate water depth, do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Sample Handling and Storage

* Following collection, place the sample in a cooler and maintain on ice.
* Samples must be delivered to the testing laboratory within 6 hours of collection or filtered and all filters frozen on dry ice within 6 hours of collection.
* In addition to collecting the sample, look for signs of disturbance throughout the reach that would contribute to the presence of fecal contamination to the water body. Record these disturbances on the Field Data Form. If samples are not delivered within 6 hours to the laboratory, the samples will be filtered. See “Method: Filtering *E. coli* Samples.”

#### Method: Filtering E. Coli Samples

If the water samples for *E. coli* will not be delivered to the testing laboratory within 4 hours, two separate filters for the *E. coli* sample need to be processed. All the filters required for an individual site should be sealed in plastic bags until use to avoid external sources of contamination and stored on dry ice.

Supply list for processing sample:

* Nitrile gloves
* Sterile screw-cap 50 mL
* Filtration apparatus with collection flask
* Sterile filter holder, Nalgene 145/147
* Vacuum pump (electric pump may be used if available)
* Sterile phosphate buffered saline
* Osmotics 47 mm polycarbonate sterile filters
* Sterile disposable forceps
* Petri dishes (60 × 15, disposable)
* Two sterile microcentrifuge tubes containing sterile glass beads
* One additional sterile microcentrifuge tube if collecting filter blank
* Bubble bag
* Zip-top bag
* Dry ice
* Cooler

For recording measurements:

* Sample collection form
* Pencils
* Fecal indicator sample labels (two vial labels and one bag label)
* Filter blank label if collecting filter blank

The sample must be filtered and frozen within 6 hours of collection.

1. Put on nitrile gloves.
2. Set up sample filtration apparatus on flat surface and attach vacuum pump. Set out 50 mL sterile PP tube, sterile 60 mm Petri dish, two bottles of chilled phosphate buffered saline, the Osmotics 47 mm polycarbonate sterile filter box, and two filter forceps.
3. Chill filter extraction tubes with beads on dry ice.
4. Aseptically transfer two polycarbonate filters from the filter box to base of opened Petri dish. Close filter box and set it aside.
5. Remove the pre-loaded cellulose nitrate filter from funnel and discard. Be sure to leave the support pad in the filter funnel.
6. Load filtration funnel with a sterile polycarbonate filter on support pad (shiny side up).
7. Shake sample bottle(s) 25 times to mix well.
8. Measure 25 mL of the mixed water sample in the sterile graduated sterile PP tube and pour into filter funnel.
9. Replace cover on filter funnel and pump to generate a vacuum (do not generate more than 7 in. of mercury of vacuum [3.44 psig]). Keep pumping until all liquid is in filtrate collection flask.
10. If the first 25 mL volume passes readily through the filter, add another 25 mL and continue filtration.
11. If the filter clogs before completely filtering the first or second 25 mL volume, discard the filter and repeat the filtration using a lesser volume.
12. Pour approximately 10 mL of the chilled phosphate buffered saline into the graduated PP tube used for the sample. Cap the tube and shake it five times. Remove the cap and pour rinsate into filter funnel to rinse filter.
13. Filter the rinsate and repeat with another 10 mL of phosphate buffered saline.
14. Remove filter funnel from base without disturbing filter. Using sterile disposable forceps, remove the filter (touching only the filter edges) and fold it in half, in quarters, in eighths, and then in sixteenths (filter will be folded four times).
15. Insert filter into chilled filter extraction tube (with beads). Filter should be inserted open end down (pointed side up) into the tube. Replace and tighten the screw cap.
16. Record the volume of sample filtered through the filter on the outer bag label and apply the label to the bubble bag. (**Do not** cover with clear tape.)
17. Insert tube(s) into bubble bag and zip-top bag on dry ice for preservation during transport and shipping.
18. Record the volume of water sample filtered through each filter and the volume of buffer rinsate each filter was rinsed with on the sample collection form, side 2. Record the filtration start time and finish time for the sample as well as the time the filters were frozen.
19. Repeat steps 6 to 15 for the remaining 50 mL sub-sample volume to be filtered. Make every effort to filter the same volume of sample through each of the two filters.

#### Pre-sampling Site Assessment

Visually scan station for best wade-in sampling site that will provide least disturbance of substrate and provide for a representative sample. Note any site conditions that may affect samples. If no water in the stream, record as “No Flow.” Where wading into the flow is not possible for the chosen site and no alternative is available, ensure that a sample taken from the bank is in the flow and representative of the larger area. If a representative location cannot be found, do not take sample.

#### Field Method: General Bottle Sampling Procedure

##### Wadeable Rivers and Streams

1. Wade in carefully, moving from downstream to upstream until you get to the main flow of the stream. Sample from midstream if the stream is small. If the stream is larger, go only as far out from shore as is safe. Establish a solid footing before filling a sample.
2. Stand facing upstream (with the water moving toward you). Stand still for a few seconds to allow any stirred-up sediment to be carried away by the current.
3. Take the readings at 0.5 m depth. Put on shoulder- or elbow-length double polyethylene sampling gloves or other skin-protective gloves (highly recommended).
4. Facing upstream, take samples directly into sample bottles. Minimize air contact with open sample bottle. Rinse inside of new, pre-cleaned sample bottles.
5. Hold the base of the container and gently plunge the capped container beneath the surface. Turn container until neck points slightly upward and mouth is directed toward the current. If there is no current, create a current artificially by pushing container forward horizontally in a direction away from your hand. Open sampling container about 6 in. below the surface (if possible) to avoid collecting surface scum. Fill container close to the neck of the container, leaving enough air space to allow for mixing. Cap underwater and wade out.
6. Place on ice to 4°C immediately.

Notes:

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples. (The person sampling must be stable while sampling, keeping feet still.)
* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Non-wadeable Rivers and Streams

If it is not safe to wade (too fast-flowing or too deep) or the stream bottom is too muddy to allow for collection of a clean sample by wading, use a sample collection rod as described below.

1. Rinse the clamp end of the rod in the stream you wish to sample. This will reduce the possibility of contamination from the previous station.
2. Place the bottle (without preservative) in the clamp and squeeze the clamp closed. Remove the cap from the bottle.
3. Rotate the rod until the bottle is upside down. Immerse the bottle to the desired depth, then rotate the rod to fill the bottle.
4. Once the bottle is full, remove from the water and pour into a bottle with preservative in it. Refill the bottle (without preservative), remove it from the water, cap it and remove it from the clamp.
5. Place on ice to 4°C immediately.
6. To collect a field blank sample: Rinse the clamp three times by pouring a small amount of distilled water over the clamp. Attach the sample bottle to the clamp. Take the blank sample by pouring an appropriate amount of distilled water into the sample bottle. Store the sample in the cooler.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).

##### Elevated Drop/Bridge Grabs (Rivers and Streams Deeper than 18 Inches at Drop Location)

If the site is not safe for either wading or using the sample collection rod, use the basket sampling method from a bridge as described below. Visually scan the drop location for unobstructed vertical drop and for approximate water depth of 18 in., and then proceed as follows from the upstream side of the drop/bridge:

1. Rinse basket three times in water to be sampled, without sample bottles in it but with any sand/water-filled weight bottle(s). Place required weight in basket as appropriate.
2. Fit the weighted basket sampler with 500 mL pre-cleaned bottle(s). Bottles should not contain any preservative.
3. Secure bottles inside basket. Do not deploy unit unless bottles are tightly secured inside basket; if not secured, bottles will pop out on entry.
4. Unscrew caps and place in a new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface, and gently plunge into water to approximately 6 in. below water surface. Allow the bottles to fill. Observe the sampler closely to ensure that it does not touch bottom sediments.
6. When bubbling stops (about 30 seconds), pull up basket slowly. While pulling the basket up, be sure no debris from the bridge or the tow line/rope is falling into the sample bottles. Pour into bottles with preservative (if needed).
7. Place on ice to 4°C immediately.

Notes:

* For analytes requiring glass, attempt to find a wade-in station to take the sample directly, or rig the basket sampler to take and protect a glass container.
* For analytes requiring a sterile plastic container, place new, pre-cleaned, sterile, 500 mL HDPE bottle in the basket, mark its location in the basket, take sample, cap it, and deliver on ice—or transfer the sterile, 500 mL sample to a smaller, sterile plastic bottle for delivery to laboratory (or attempt to find a wade-in station to take the sample directly).
* If water depth is less than approximately 18 in., do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

#### Field Method: Collecting Duplicates and Field Blanks

1. To collect a duplicate sample, repeat the steps above exactly for each bottle. Collect the duplicate right after the first sample. Duplicate and blank sample bottles should be pre-labeled accordingly.
2. To collect a blank sample, pour distilled water from the bottle of distilled water directly into the sample bottle.

+++END-IF+++

## B3 Sample Handling and Custody

Labels with the following information will be attached to sample containers:

* Sample number
* Site ID
* Time and date of collection
* Preservation requirements
* Name of sampler and organization

Samples for shipment will be prepared as follows:

* All samples will be appropriately preserved and packaged for transport.
* If obtainable samples are missing, the Project Manager and Field Coordinator will determine corrective action (e.g., reschedule a site visit or return to the site that same day to complete collection of the missing samples).
* All samples will be labeled and the labels checked for completeness, legibility, accuracy, and consistency.
* Labels and forms will be reviewed to ensure consistent sample ID information.
* Each sample container will be inspected to make sure there are no leaks and that all containers are properly sealed.

The Field Coordinator will complete the chain of custody form(s) for samples shipped to a laboratory. A copy of tracking forms will be made and retained by the team. The original form will be sent in the container with the sample. Copies of all tracking forms will be included in the coolers when the Field Coordinator sends samples to the labs.

## B4 Analytical Methods

## In situ parameters measured by calibrated sensors on site, including temperature, dissolved oxygen, conductivity, pH and turbidity, do not require analytical methods. Laboratory analysis of discrete samples collected in the field will be conducted as indicated.

Table B4.1. Approved Analytical Methods

| Parameter - Method | MDL (mg/l unless stated) | Lab Name |
| --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality')+++ |  |  |
| +++ **INS $**parameter.label+++ | +++ **INS $**parameter.mdl+++ |  |
| +++END-FOR parameter +++ |  |  |

## B5 Field and Analytical Laboratory Quality Control

### The monitoring project will include appropriate field and laboratory QC samples to assess general data quality issues, as well as specific data quality objectives. Field QC samples will be taken for 10% of all water quality samples taken per sampling event.

### B5.1 Field Duplicates

Duplicates will be taken side by side and simultaneously. Field duplicates are submitted to the laboratory along with all other samples.

### B5.2 Field Blanks

Ambient field blanks will be taken at 10% of total samples to evaluate if sample contamination may have occurred due to improper sample collection, atmospheric fallout, or other causes. A field blank will be created by filling a clean sample bottle with de-ionized or distilled water in the field during sampling activities, then treated the same as other samples taken from the field (i.e., labeled, stored on wet ice in cooler). Field blanks are submitted to the laboratory along with all other samples and are used to detect any contaminants that may be introduced during sample collection, fixing, storage, analysis, and transport.

Table B5.1. Quality Control Measures

| Parameter - Method | Accuracy Checks | Precision Checks | % Field QC Samples |
| --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality') +++ |  |  |  |
| +++ **INS $**parameter.label+++ | +++ **INS $**parameter.accuracyChecks+++ | +++ **INS $**parameter.precisionChecks+++ | +++ **INS $**parameter.percentFieldQC+++ |
| +++END-FOR parameter +++ |  |  |  |

Table B5.2. Field Quality Control: ￼ (measured using sensors)

| Parameter | Check Description | Frequency | Acceptance Criteria | Corrective Actions |
| --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality' && (param.method === 'in situ' || param.method === ''))+++ |  |  |  |  |
| +++ **INS $**parameter.parameter+++ |  | +++ **INS $**parameter.frequency+++ |  | +++ **INS $**parameter.correctiveAction+++ |
| +++END-FOR parameter +++ |  |  |  |  |

+++IF determine('Freshwater Water Quality', 'Freshwater','Total phosphorus', '') === true || determine('Freshwater Water Quality', 'Freshwater',' Total nitrogen', '') === true || determine('Freshwater Water Quality', 'Freshwater',' Ammonia-N', '') === true || determine('Freshwater Water Quality', 'Freshwater','Nitrate-Nitrite-N', '') === true || determine('Freshwater Water Quality', 'Freshwater','Orthophosphate', '') === true+++

Table B5.3. Field Quality Control: Nutrients

| Quality Control Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Water Chemistry Container and Preparation | Rinse collection bottles 3xwith ambient water before collecting water samples. | Discard sample. Rinse bottle and refill. |
| Sample Storage | Store samples in darkness at 4°C. Confirm cooler/sample temperature on delivery to lab (e.g. use temperature blank).  Deliver to laboratory within hold time. | Qualify sample as suspect for all analyses. |

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Chlorophyll-a', '') === true+++

Table B5.4. Field Quality Control: Chlorophyll *a*

| Quality Control Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Chlorophyll-a Containers and Preparation | Rinse collection bottles 3x with ambient water before collecting water samples. | Discard sample. Rinse bottle and refill |
| Holding Time | 24 hours | Qualify samples |
| Sample Storage | Chl a samples are shipped on wet ice | Qualify sample as suspect |

+++END-IF+++

Table B5.5. Data Validation Quality Control for Water Chemistry

| Activity | Requirements and Corrective Action |
| --- | --- |
| Range checks, summary statistics, and/or exploratory data analysis | Current reporting errors or qualify as suspect of invalid |
| Review holding times | Qualify value for additional reviews |
| Review data from QA samples | Determine impact and possible limitations on overall data usability |

+++IF determine('Freshwater Water Quality', 'Freshwater', 'E. coli', '') === true+++

Table B5.6. Field Quality Control: Fecal Indicator

| Quality Control Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Check integrity of sample containers and labels | Clean, intact containers and labels | Obtain replacement supplies |
| Sterility of sample containers | Sample collection bottle and filtering apparatus are sterile and must be unopened prior to sampling. Nitrile gloves must be worn during sampling and filtering | Discard sample and recollect in the field. |
| Sample Collection | Collect sample at the last transect to minimize holding time before filtering and freezing | Discard sample and recollect in the field. |
| Sample holding | Sample is held in a cooler on wet ice until filtering. | Discard sample and recollect in the field. |
| Field Processing | Sample is filtered within 6 hours of collection and placed on dry ice. | Discard sample and recollect in the field |
| Field Blanks | Field blanks must be filtered at 10% of sites. | Review blank data and flag sample data. |

Table B5.7. Data Validation Quality Control: *E. coli*

| Check Description | Frequency | Acceptance Criteria | Corrective Action |
| --- | --- | --- | --- |
| Duplicate sampling | Duplicate composite samples collected at 10% of sites | Measurements should be within 10 percent | Review data for reasonableness; determine if acceptance criteria need to be modified |
| Field filter blanks | Field blanks filtered at 10% of sites | Measurements should be within 10 percent | Review data for reasonableness; determine if acceptance criteria need to be modified |

+++END-IF+++

+++IF determine('Freshwater Water Quality', 'Freshwater', 'Microcystins', '') === true+++

Table B5.8. Field Quality Control: Microcystins

| Quality Control Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Holding time | Hold sample on wet ice and freeze immediately upon return to base. Keep frozen until shipping | Quality samples |
| Sample storage | Store samples in darkness and frozen (-200C)  Monitor temperature daily | Qualify samples as suspect |

Table B5.9. Data Validation Quality Control: Microcystins

| Activity or Procedure | Requirements and Corrective Action |
| --- | --- |
| Range checks, summary statistics, and/or exploratory data analysis | Current reporting errors or qualify as suspect of invalid |
| Review holding times | Qualify value for additional reviews |
| Review data from QA samples | Determine impact and possible limitations on overall data usability |

+++END-IF+++

## B6 Instrument/Equipment Inspection and Testing

All equipment used to collect or analyze ambient or collected samples will undergo periodic maintenance and calibration verification performed by manufacturer’s representatives or service consultants. These procedures will be documented by date and the signature of person performing the inspection. (For example, multi-parameter probes will receive annual (or as needed) maintenance and calibration checks by manufacturers or certified service centers.) All other sampling gear and laboratory instrumentation will be maintained in good repair as per manufacturer’s recommendations to ensure proper function.

Records of equipment inspection, maintenance, repair, and replacement will be kept in a logbook, along with standard operating procedures for instrument maintenance and calibration.

Table B6.1. Typical Instrument/Equipment Inspection and Testing Procedures

| Parameter - Method | Equipment | Inspection frequency | Type inspection | Maintenance, Corrective Action | Person Responsible |
| --- | --- | --- | --- | --- | --- |
| Total N, Nitrate-Nitrite-N, Ammonium-N, Total P, Orthophosphates, chlorophyll –a, chlorides, total suspended solids, E. colimicrocystins | Sample bottles | Before each use | Visual for integrity, cleanliness | Acid washed prior to use (or clean-certified from manufacturer or lab) |  |
| Total N, Nitrate-Nitrite-N, Ammonium-N, Total P, Orthophosphates, chlorophyll –a, E. coli, microcystins | Filtering apparatus | Before each use | Proper functioning, clean storage | Spare filters, syringe |  |
| Temperature, conductivity, DO, pH, turbidity | Meters | Before each use | Battery life, DO membrane | Spare batteries, spare membranes |  |
| Geographical coordinates | GPS | Before each use | Battery life | Repair, replace, spare batteries on hand. |  |

## B7 Field Equipment/Maintenance, Inspection, and Calibration

### B7.1 Pre-measurement Instrument Checks and Calibration

Field instruments will be tested and calibrated prior to sampling, either prior to departure for the site or at the site.

Site location will be verified using a GPS receiver. Field crews will have access to backup instruments if any instruments fail the manufacturer performance tests or calibrations. Prior to departure, the following checks and calibrations will be performed:

* If using a handheld GPS unit, turn on the GPS receiver and check the batteries. Replace batteries immediately if a battery warning is displayed.
* Test and calibrate the multi-parameter sensor (or sonde) according to the manufacturer's calibration and maintenance procedures. Records of these checks should be saved in a logbook or other documentation.

#### Multi-Parameter sensor

The dissolved oxygen, pH, temperature, and conductivity sensor functions of the multi-parameter sensor or individual sensors will be calibrated prior to departure to the sampling site(s). A single calibration is sufficient for the day.

Table B7.1. Instrument Calibration Procedures

|  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- |
| Parameter | Instrument | Type of Inspection | Inspection and Calibration Frequency | Standard of Calibration Used | Corrective Action |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Freshwater Water Quality') +++ |  |  |  |  |  |
| +++ INS $parameter.parameter+++ | +++ INS $parameter.instrument+++ |  | +++ INS $parameter.calibrationFrequency+++ | +++ INS $parameter.calibrationStandard+++ | +++ INS $parameter.correctiveAction+++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

“External standards” refers to standards of reliable quality obtained from reputable commercial or other suppliers; “known standards” refers to those where the value is known before calibration.

### B7.2 Post-measurement Calibration Check—Multi-Parameter sensor

After all sensor measurements have been completed for the sampling day, a post-measurement calibration check of the parameter sensor is performed. To do this, pH, conductivity, and DO of one of each of the respective calibration standards that were used earlier in the day to calibrate the instrument will be measured and values recorded. If significant drift is detected (as defined the manufacturer), the sensor may need service; data collected since the last successful calibration and post-measurement calibration check will be flagged.

### B7.3 Instrument/Equipment Inspection, Testing Procedures

Equipment maintenance will be conducted routinely. Records of equipment inspection, maintenance, repair, and replacement will be recorded in a logbook.

## B8Inspection/Acceptance of Supplies and Consumables

The Project Manager will be responsible for ensuring correct sample handling by:

* Ensuring availability of all required sampling supplies in the field.
* Properly labeling all sample containers for biological samples in the field.
* Recording all relevant sampling information on the respective field forms and chain-of- custody forms.
* Coordinating the transfer of all samples from the field to laboratories for analysis.

Table B8.1. Critical Field Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

|  |  |  |
| --- | --- | --- |
| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| Jars for macrofaunal samples | Visually inspected for cracks, breakage, and cleanliness. May be reused. |  |
| 95% ethanol | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment | Visually inspected for obvious defects, damage, and contamination. |  |
| Navigation instruments, digital camera | Functional checks to ensure proper calibration and operating capacity. |  |

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other studies water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records.
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments).

Secondary data used will be documented in the Secondary Data Form, attached, according to Sections A9 and C2.

~~To verify that any data used are of known and documented quality and are consistent with project data quality objective, the following metadata will be provided for each data source:~~

* ~~Title of document~~
* ~~Source of information~~
* ~~Notes on quality of data, including whether quality is demonstrated through a QAPP or some other means~~
* ~~As applicable, a statement on planned restrictions on use of the data due to questions about data quality~~

~~Table B9.~~1~~. Examples of Secondary Data Providers~~

| ~~Data source~~ | ~~Waterbody type~~ | ~~Sample data parameters~~ | ~~Sample design~~ | ~~Geographic area~~ | ~~Web data links~~ |
| --- | --- | --- | --- | --- | --- |
| ~~DCR~~ | ~~Lakes & ponds~~ | ~~Secchi depth~~  ~~Nutrients~~  ~~Chlorophyll a~~  ~~Bacteria~~  ~~Non-native plants~~ | ~~Targeted~~ | ~~MassBays wide~~ | ~~Website URL~~ |
| ~~USGS~~ | ~~Rivers & streams~~ | ~~Streamflow~~ | ~~NA~~ | ~~National~~ |  |
| ~~NWS~~ | ~~All~~ | ~~Weather data~~ | ~~NA~~ | ~~National~~ |  |

## B10 Data Management

Field crew shall record sampling data on Field Data Forms, review them, sign them, and submit them to the Project Field Coordinator. The Project Field Coordinator will review the forms and confer with the crew on any required corrective action. Field crew will fill out the chain of custody form for forwarding the samples (processed or unprocessed) to the laboratory. Each person who handles or transports samples will also sign the custody form upon receipt of samples. Chain of custody forms will accompany samples to the lab and be returned to the Monitoring Program Coordinator after each analysis run is completed.

Once laboratory analyses are complete, the laboratory personnel will email or mail laboratory results to the Monitoring Program Coordinator. The Monitoring Program Coordinator and/or Data Management Coordinator will enter raw field and lab data electronically. Data are then compared with field sheets for accuracy. The original Field Data Forms will be stored in the organization’s office. Electronic backups and copies of Field Data Forms will be made and stored.

Data quality control steps will be taken at several stages. Documentation of data recording and handling, including all problems and corrective actions, shall be included in all preliminary and final reports. (Corrective action reporting form attached.) See Section A9 for recording handling and storage procedures.

~~Table B10.~~1~~. Table 10.1. Data Management, Review, Validation, and Verification Process~~

| ~~Activity~~ | ~~By whom~~ | ~~Corrective action, if needed~~ |
| --- | --- | --- |
| ~~Check labels prior to sampling to ensure correct labeling of container~~ | ~~Field sampler~~ | ~~Correct label or change container~~ |
|  |  |  |
|  |  |  |

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' , '', '') === true+++

# Section B. Fresh Water/Benthic Data Generation and Acquisition

## B1 Sampling Design

Ideally, site selection criteria will be clear, ~~concise~~ so that if two researchers were to follow them, each would choose similar locations. The criteria should minimize the amount of subjectivity that enters into the site selection process. If a reference site is used, the conditions at reference sites should represent the best range of minimally impaired conditions that can be achieved by similar streams within a particular ecological region (Hughes et al. 1995).

+++IF determineConcern('Eutrophication (Nutrients)') === true || determineConcern('Illicit Connections') === true || determineConcern('Stormwater') === true +++

### B1.1 Sampling Site Selection

For point source assessments, the sampling site selection should include stations upstream and downstream of the source, as well as at least three regional reference stations. The site selected should be in an area where reasonable opportunity for mixing of the effluent has occurred. If a mixing zone has been defined in a license, sampling should occur immediately downstream of it. In cases where the effluent plume channels down one bank for great distances (over 1 km), or where localized effluent impact is expected to be severe for a distance beyond the zone of initial dilution, it is advisable to have a sampling site upstream of the source, one or more in the plume, and at least two farther downstream. One downstream site should be located at the point of presumed bank to-bank mixing and subsequent sites should be located to assess the extent of impact downstream. The area of initial dilution of an effluent should be determined by visual observation of the plume pattern; by observations of biotic effects attributable to the plume, if evident (benthic algal biomass growth, die-off patterns); and by transects of specific conductance measurements from the outfall, in a downstream direction.

Selection of sampling sites should ensure that the physical characteristics among sampling sites are similar. If surveys are conducted to determine use designations, sampling locations should be representative of the stream reach. Reference conditions should include minimally impaired sites in the same ecoregion, size class, and stream type (width, depth, gradient).

+++END-IF+++

+++IF determineConcern('General Environmental Health: Physical/Chemical Water Quality') === true || determineConcern('Recreation (Swimming and/or Boating)') === true || determineConcern('Harmful Algal Blooms (HABs) (Algal toxins)') === true || determineConcern('General Environmental Health: Benthic') === true || determineConcern('Acidification') === true +++

### B1.2 Sampling Site (Reach) Selection and Assessment

For biological and habitat assessment, sampling should provide a representative picture of the ecological community. Unless basically comparable physical habitat is sampled at all stations, community differences attributable to habitat degradation will be difficult to separate from those resulting from water quality degradation (Plafkin et al. 1989). Sampling highly similar habitats will also reduce metric variability, attributable to factors such as current speed and substrate type.

To meaningfully evaluate biological condition, sampling locations must be carefully selected to ensure generally comparable physical habitat. Sample locations should be scouted out ahead of time to identify appropriate reaches and to determine substrates that are available for sampling. Station siting of both study sites and reference (regional and/or site-specific) sites ideally takes place during June. Reconnaissance activities prior to June are not desirable, as instream conditions—most notably flow regimes—during this time are often dramatically different than during the sampling index period. To lay out the sampling reach and sites, first make sure the route to the site is free of obstacles that would prohibit sampling and data collection activities, then assess the sample reach characteristics. Field conditions (e.g., instream and riparian habitat characteristics, surrounding land use, observations of nonpoint source pollution or other pertinent information) during the time of reconnaissance will be noted and recorded in a field notebook.

+++END-IF+++

## B2 Sampling Methods – all freshwater benthic parameters

### B2.1 Site Photographs

At all sample reaches, photographs—at least one upstream and one downstream—will be taken with a digital camera. These and any additional photos will be recorded in the Photo log (sample attached) with a brief description.

### B2.2 Flow Velocity

Flow velocity will be determined using the float method, as described below.

#### Equipment

* Ball of heavy-duty string, four stakes, and a hammer to drive the stakes into the ground
* Tape measure (at least 20 ft)
* Orange
* Net
* Stopwatch
* Calculator

#### Method

1. Measure off 25 ft along the bank of a straight section of stream. To measure discharge, choose a stretch of stream that is straight (no bends), at least 6 in. deep, and without an area of slow water such as a pool. Unobstructed riffles or runs are ideal. Stretch string across each end of the 25 ft length.
2. Release the orange at the upstream site, positioned so that it flows into the fastest current. Using a stopwatch, record the time it takes to flow from the upstream transect line to fully pass under the downstream transect line. (If the float moves too fast for an accurate measurement, measure off 50 or 75 ft instead of 25.) Repeat the measurement at least two more times, more for greater accuracy. Average the results (in seconds).
3. Calculate flow velocity as distance traveled (ft) divided by the average amount of time (seconds) it took the orange to travel that distance.

+++IF determine('Freshwater Benthic', 'Freshwater' , 'Benthic algal biomass', '') === true+++

## B2 Sampling Method—Viewing Bucket

Semi-quantitative assessments of benthic algal biomass and taxonomic composition are made using a viewing bucket (marked with a grid) and a biomass scoring system. The advantage of using this technique is that it enables rapid assessment of algal biomass over larger spatial scales than substrate sampling and laboratory analysis. Coarse-level taxonomic characterization of communities is also possible with this technique. This technique (developed by R.J. Stevenson and S.T. Rier [reuse EPA 1999 footnote here]) is a survey of the natural substrate and requires no laboratory processing, but hand-picked samples can be returned to the laboratory to quickly verify identification.

### B2.1 Equipment

* Meter stick
* Pencil
* Algal biomass Field Data Form
* Viewing bucket with 50-dot grid

Make the viewing bucket by cutting a hole in bottom of large (0.5 m diameter) plastic bucket, but leave a small ridge around the edge. Attach a piece of clear acrylic sheet to the bottom of the bucket with small screws and silicon caulk. (The latter makes a watertight seal so that no water enters the bucket when it is partially submerged.) Mark 35 dots in a seven-by-five grid on the top surface of the acrylic sheet with a waterproof black marker or clear divot in the acrylic (Figure B2.2).

### B2.2 Procedure

1. Identify three transects and select three locations along each transect, one near the right bank, one near the middle, and one near the left bank (looking downstream). Focus on the “most productive” sites along those transects, generally riffles, runs, or snags. Avoid pools or rapids.
2. Identify one person to conduct the visual survey (viewer) and one person to fill out the field sheet (recorder) (Figure B2.1).
3. At each location, the recorder notes the transect, and sample number, and instantaneous dissolved oxygen, temperature, pH, and flow velocity using field sensors.
4. The viewer then immerses the viewing bucket in the water (Figure B2.2). Benthic algae can be clearly viewed by looking down through the bucket when it is partially submerged in the stream. To minimize glare, it is sometimes helpful to put a little water inside the viewing bucket.

|  |  |
| --- | --- |
|  |  |
| Figure B2.1. Using the viewing bucket, recorder notes observations relayed by the viewer. | Figure B2.2. Viewing bucket for qualitative benthic algae assessment. |

1. While viewing through the bucket, identify points on the stream bottom below the upper left dot and the lower right dot to help keep the bucket in the same area. Take a digital photograph of a card with the sample site ID, water body name, and date written in large, thick letters, followed by a photograph of the viewing area.
2. To characterize macroalgal biomass:
3. Observe the bottom of the stream through the bottom of the viewing bucket and count the number of dots that appear over macroalgae (e.g., *Cladophora* or *Spirogyra*) under which substrates cannot be seen. Record the number of dots and the type of macroalgae under those dots on the Field Data Form.
4. Measure and record the maximum length of the macroalgae.
5. If two or more types of macroalgae are present, count the dots, measure, and record information for each type of macroalgae separately.
6. If there is a mixture of decomposing filaments, microalgae, and silt, then treat it as being a mat type (Step 8) and not a filament.
7. To characterize extent of suitable substrate:
   1. Record the number of dots under which there is gravel greater than 2 cm in diameter.
8. To characterize microalgal cover:
9. The viewer will begin from the upper left dot and work across each row, calling out the categories under 8.b and 8.c for the recorder to document on the Field Data Form. The recorder will make a hash mark under the corresponding category.
10. Determine the kind (usually diatoms and blue-green algae) and estimate (or measure with the ruler) the thickness (density) of microalgae under each dot using the following thickness scale:

* Mat 0—substrate is rough or slightly slimy with no visible algae.
* Mat 1—a thin layer of microalgae is visually evident; underlying rock is still visible.
* Mat 2—accumulation of microalgal layer from 0.5–1 mm thick is evident; underlying rock is covered.
* Mat 3—accumulation of microalgae layer from 1–5 mm thick is evident.
* Mat 4—accumulation of microalgal layer from 5 mm to 2 cm thick is evident.
* Mat 5—accumulation of microalgal layer greater than 2 cm thick is evident.

1. Record previously unidentified growth under each dot using the following labels:

* Sand/clay/mud
* Plant—an aquatic plant or plant-like macroalga (*Batrachospermum* or *Lemmanea*)
* Moss
* Crust—a crust-forming algae (may be black, red, or green)
* Sewage fungus—a filamentous bacteria, such as *Sphaerotilus*. Does not include iron or manganese bacteria. Try to bring a sample back for verification.
* Sponge—a freshwater sponge

1. Before the viewer moves the viewing bucket, the recorder will:
2. Convert the number of hash marks for each category to a numeral and record that number under the category.
3. Add up the number of hash marks in the row to make sure that 35 observations were taken for statistical significance. If not, then the viewer can make additional observations or subtract the most recent observations to get a total of 35 observations. It is acceptable to have more than 35 observations if it is not clear what to remove.

## B3 Sample Handling—Algal Biomass

Filamentous algae not able to be identified in the field will be subsampled, transferred to a plastic bag with clean stream water, and labeled with the sample number, site ID, and name of sampler. The algal sample will be held on ice for transport to the sample analysis site hours.

## B4 Analytical Methods—Algal Biomass

Any algae collected will be observed under a dissecting microscope within 72 hours of collection for identification to the genus level.

Density of algae on substrate may be determined using the following statistics:[[7]](#footnote-8)

1. Maximum length of each type of macroalgae.
2. Maximum density of each type of microalgae on suitable substrate (i.e., categories Mat 0 through Mat 5 as described in Section B2).
3. Average percent cover of the habitat by each type of macroalgae:

% cover = 100 × *Dm/Dt*

*Dt* = total number of grid points (dots) evaluated at the site

*Dm* = number of grid points (dots) over macroalgae

1. Mean density (i.e., thickness rank) of each type of macroalgae on suitable substrate (listed in Section B2 under categories Mat 0 to Mat 5):

mean density = ∑*diri/dt*

*dt* = total number of grid points (dots) over suitable substrate for microalgae at the site

*di* = number of grid points over microalga of different thickness ranks for each type of microalga

*ri* = thickness rank

+++END-IF+++

1. +++IF determine('Freshwater Benthic', 'Freshwater' , 'Macroinvertebrates', 'Kick sampling') === true+++

## B2 Sampling Methods—Kick Sampling

### B2.1 Method Summary

Benthic macroinvertebrate samples will be collected using a net with 500 µm mesh openings. Samples will be preserved in the field with 95% ethanol, then sorted and sent to the laboratory for identification.

### B2.2 Equipment and Supplies

* Net, 500 µm mesh
* Timer or stopwatch
* Soft nylon brush
* Forceps
* Small spatula, spoon, or scoop to transfer sample
* Sample jars (1 L HDPE plastic suitable for use with 95% ethanol)
* 95% ethanol, properly stored and labeled
* Wash bottle (1 L, labeled “stream water”)
* Cooler (with absorbent material for transporting ethanol and samples)
* Plastic electric tape
* Scissors
* Blank and completed labels
* Indelible-ink markers (for labels)
* Pencils
* Clear tape
* Kick sample collection Field Data Form
* Blank labels on waterproof paper for internal sample labels

### B2.3 Sampling Procedure

Stream sampling site will be a wadeable, 100 m long reach representing the best available habitat in the desired river reach. Sampling points located in water that is too deep or unsafe for wading will be avoided. Ideal substrate and habitat types are:

*Gravel (G):* fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm)

*Coarse (C):* Cobble to boulder (tennis ball to car sized; 64 mm to 4000 mm)

*Pool (P):* Still water; low velocity; smooth, glassy surface; usually deep compared to other partsof the channel

*Glide (GL):* Water moving slowly, with smooth, unbroken surface; low turbulence

*Riffle (RI):* Water moving, with small ripples, waves, and eddies; waves not breaking, and surfacetension is not broken; “babbling” or “gurgling” sound

1. With the net opening facing upstream, quickly position the net securely on the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the net from seating properly on the stream bottom. If the stream is only one net wide at a transect, the net will be placed across the entire stream width.
2. Holding the net in position on the substrate, visually define a quadrat that is one net width wide and long upstream of the net opening.
3. Hold the net securely in position. Starting at the upstream end of the quadrat, vigorously kick the remaining substrate.
4. When substrates are too large or difficult to kick, organisms may be dislodged by rubbing the substrate item into the net.
5. Pull the net up out of the water. Rinse any large debris items (rocks, sticks, etc.) caught in the net and return them to the stream, once any macroinvertebrates clinging to them are removed.
6. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid letting any water or material enter the mouth of the net during this operation.
7. Ten kick samples from a square area with dimensions equal to the width of the net opening will be composited for a total area sampled of about 2 m2.
8. In streams where the riffles within the reach are inadequate to allow for a 2 m2 composite, other productive habitats may be sampled by jabs into snags or rubbing substrates. In such cases, record notes on the field sheets indicating the number of kicks or jabs in each habitat category that contributed to the composite sample.
9. Rinse any debris (gravel, rocks, sticks, etc.) caught in the net in the net and return it to the stream, once any macroinvertebrates clinging to it have been removed. Clean and remove as much gravel as possible so that organisms do not get damaged. Place the residue in the net in a container with enough denatured 95% ethanol added to cover the residue.
10. Put a label (template attached) filled in with a pencil inside the sample container, and affix a duplicate label with the words “preservative: denatured 95% ethanol” to the outside of the container.

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' , 'Macroinvertebrates', 'Rock baskets') === true+++

## B2 Sampling Method—Rock Baskets

### B2.1 Method Summary

Rock baskets will be deployed when stream substrates are not ideal for kick sampling (too deep, benthic substrates consisting of fines or ledge), or where passive collection of macroinvertebrates can provide a more complete assessment of the system. Baskets contain roofing stone of a specific size class, and are left in place and undisturbed for six to eight weeks. Composite samples from multiple sites are preserved, then sorted and sent to the laboratory for identification.

### B2.2 Equipment List

* Rock-filled wire or mesh baskets, three per sampling site
* Substrate: clean, washed cobble (1 to 3 in. diameter) or #2 roofing stone (not crushed rock)
* Basket mesh size: 1.5 to 2.5 cm
* Fill weight: 7.25 + 0.5 kg
* Sieve bucket with 500 μm mesh
* Soft nylon brush
* Forceps
* Small spatula, spoon, or scoop to transfer sample
* Sample jars (1 L HDPE plastic suitable for use with 95% ethanol)
* 95% ethanol properly stored and labeled
* Wash bottle (1 L, labeled “stream water”)
* Cooler (with absorbent material for transporting ethanol and samples)
* Plastic electric tape
* Scissors
* Blank and completed labels
* Indelible-ink markers (for labels)
* Pencils
* Clear tape
* Kick sample collection Field Data Forms
* Blank labels on waterproof paper for internal sample labels

### B2.3 Sampling Procedure

1. Select similar microhabitats (e.g., riffle, pool, glide) for replicate sampling. Baskets should be submerged for the duration of deployment, and not subject to tampering.
2. Approach the location so as to avoid any disturbance in, or upstream of, the sampled site. Orient baskets with the long axis parallel to stream flow. Provide for retrieval of baskets by flagging trees in the vicinity and/or by drawing a diagram with appropriate landmarks indicated.
3. Exposure periods are 28 days +/- four days during the sampling season. Extended exposure periods may be necessary to allow for adequate colonization in the case of assessments of low-velocity or impounded habitats. If such conditions exist, a 56-day +/- four days exposure period may be used.
4. At the completion of the exposure interval, approach from downstream to retrieve the basket(s). Retrieve each basket by pressing a kick-net tightly against the streambed along the basket’s downstream edge. Then move the basket carefully into the net before lifting it through the water column. Where the water is deep enough to make this procedure difficult or impossible, use a basket made with 500 μm netting on its bottom surface to prevent the loss of organisms through the bottom of the sampler as it is raised to the surface.
5. Remove vegetation or debris snagged on the outside of the recovered sampler, taking care to avoid jarring the sampler. Place the basket in a large sieve, bucket, or tub of water. Empty net contents into the bucket as well. Then open the sampler, empty it into the bucket, rinse it inside the bucket until it is free of any organisms or adhering material, and set it aside. Rinse each rock similarly and set it aside. Then sieve the material remaining in the bucket.
6. Wash the contents of the sieve to one side by gently agitating the sieve in the water. Transfer the residue to a labeled 1 L sample bottle using minimal stream-water wash, funnel, forceps, and spoon/spatula as needed. Add 95% ethanol equal to a final concentration no less than 70% ethanol (1:2 sample water plus collected materials to 95% ethanol).
7. Place a label filled in with pencil inside the sample container, and affix a duplicate label with the words “preservative: denatured 95% ethanol” to the outside of the container.

+++END-IF+++

+++IF determine('Freshwater Benthic', 'Freshwater' , 'Macroinvertebrates','') === true +++

## B2 Sample Processing (Sorting)

Sample processing involves separating macroinvertebrates from other materials in the sample. This can be left to the laboratory, or fauna can be sorted within 72 hours prior to delivery to the taxonomist by trained personnel. For stream biomonitoring, Massachusetts DEP considers the category “macroinvertebrate” to include:

* All aquatic *Annelida*
* All aquatic *Mollusca*
* Aquatic macro *Crustacea* (except as noted below)
* All aquatic *Arachnida*
* The aquatic life stages of *Insecta* except *Hemiptera* and adult *Coleoptera* other than *Elmidae*

Macroinvertebrates excluded from the above list are not used for one of three reasons: there is insufficient ecological information on them to make them useful for biomonitoring, they are surface film dwellers, or they are capable of escaping the aquatic environment at will to avoid temporarily unfavorable conditions. One further exception is crayfish (class *Crustacea,* family *Cambaridae*), which often are seen evacuating the immediate area as kick-sampling begins and even swimming out of the kick-net. Crayfish species are noted when present in the sample but are not counted toward total numbers.

The following “guide to picking” is provided by the Charles River Watershed Association[[8]](#footnote-9).

### B2.1 Equipment List

* 70% ethanol
* Nitrile or latex gloves
* 500 μm mesh sieve
* Two white plastic trays, one marked with a 3 in. by 3 in. grid
* 250 mL bottle (preferably with no lip)
* Sufficient light source
* Waste bucket
* Forceps
* Label tape
* Resistall label paper
* Pencils or waterproof ink pens
* Magnifying glass

### B2.2 Procedure

1. Prepare a clean jar with a label. Fill the jar with 70% ethanol.
2. Empty the contents of the field-sampled jar into 500 μm mesh sieve, then thoroughly rinse the sample to remove ethanol and fine debris.
3. Collect and dilute 90% ethanol from the field sample in a 4:1 ratio with tap water before disposal.
4. Pick out any large debris and look it over carefully to check for macroinvertebrates before discarding it.
5. Empty the remaining contents of the sieve into the first (non-marked) tray.
6. Move a small amount of the material from the tray onto the gridded tray and add a small amount of tap water so everything can be moved around easily.
7. Looking through one square of the grid at a time, pick out macroinvertebrates with forceps and put them into the labeled jar. (Do not use your bare hands.)
8. Once you have searched every square of the grid, shake the tray gently to mix up the remaining contents.
9. Again, search every square of the grid to make sure you have found all the macroinvertebrates.
10. Discard any water and debris remaining on the tray.
11. Move the next portion of the sample onto the gridded tray and search it.
12. Once you have finished picking through the entire sample, make sure the contents of the labeled jar are completely covered in 70% ethanol.
13. Copy out the identification code from the jar’s label onto waterproof paper using a pencil.
14. Put this new label inside the jar and close the jar securely.
15. Rinse the original sample jar for reuse in future sampling.
16. Store all full jars at room temperature in a secure location.

## B3 Sample Handling and Custody

Macroinvertebrate samples (stored in sturdy coolers) will be delivered by a survey crew member to the contracted laboratory. A crew member will contact laboratory staff to arrange a time for sample dropoff. This will allow laboratory staff to be prepared for sample receipt. The samples, while still in approximately 70% ethanol, can be shipped by ground (or two-day express delivery if necessary) for delivery to the contracted laboratory. The lids on the sample jars will be taped and the jars inserted individually into large zip-locked or tied plastic bags lined with absorbent padding.

## B4 Analytical Methods

Analytical methods for macroinvertebrate identification will be carried out by a taxonomist according to Massachusetts DEP document CN 226.0, Section 12 (available at <https://www.mass.gov/guides/water-quality-monitoring-quality-management-program>; listed incorrectly as CN 266.0 as of August 1, 2019). Specimens will be identified to genus or species as allowed by available keys, specimen condition, and specimen maturity. Based on the taxonomy various community, population, and functional parameters, or “metrics,” are calculated, which allow an investigator to measure important aspects of the biological integrity of the community.

## B5 Field Quality Control

### B5.1 Field Sampling Quality Control

The table in this section summarizes field quality control. Duplicate samples will be collected at 10% of the stations sampled for each watershed, two samples are collected “side by side”: that is, a second kick sample or adjacent rock basket (i.e., the duplicate) is taken adjacent to one of the samples collected along one of the 10 transects sampled for each reach. Duplicate samples are preserved in a separate sample bottle marked “duplicate” and with all other information regarding station location. Duplicate samples will be used in the calculation of precision of the benthos data.

Table B5.1. Macroinvertebrate Sampling Quality Control

|  |  |  |  |
| --- | --- | --- | --- |
| Field QC | Frequency Number | Corrective Action (CA) | Persons Responsible for CA |
| Post-sampling rinse, inspection, and pick of nets, sieves, and pans | At all stations | Discard sample and re-sample if not performed | Macroinvertebrate Survey/Sample Coordinators |
| Pre-sampling rinse, inspection, and pick of nets, sieves, and pans | At all stations | Discard sample and re-sample if not performed | Macroinvertebrate Survey/Sample Coordinators |
| On-site sample preservation (95% ethanol-macroinvertebrates) | All macroinvertebrate samples | Preserve at lab within or discard | Macroinvertebrate Survey/Sample Coordinators |
| Collection of duplicate samples at various stations to assess the consistency of the collection effort | 10% of total number of samples collected for each watershed | Re-sample if not performed | Macroinvertebrate Survey/Sample Coordinators |

Sample labels must be properly completed, including the sample identification code, date, stream name, sampling location, and collector’s name, and placed into the sample container. The outside of the container should be labeled with the same information. Chain of custody forms, if needed, must include the same information as the sample container labels.

After sampling has been completed at a given site, all nets, pans, etc., that have come in contact with the sample should be rinsed thoroughly, examined carefully, and picked free of organisms or debris. Any additional organisms found should be placed into the sample containers. The equipment should be examined again prior to use at the next sampling site.

### B5.2 Quality Control for Sorting/Picking

Ten percent of the sorted samples in each lot should be examined by laboratory QC personnel or a qualified coworker. (A lot is defined as a special study, basin study, entire index period, or individual sorter.) The QC worker will examine the grids chosen and tray used for sorting and will look for organisms missed by the sorter. Organisms found will be added to the sample vials.

If the QC worker finds fewer than 10 organisms (or 10% in larger subsamples) remaining in the grids or sorting tray, the sample passes; if more than 10 (or 10%) are found, the sample fails. If the first 10% of the sample lot fails, the QC worker will check a second 10% of the sample lot. Sorters in-training will have their samples 100% checked until the trainer decides that training is complete.

After processing is complete for a given sample, all sieves, pans, trays, etc., that have come in contact with the sample will be rinsed thoroughly, examined carefully, and picked free of organisms or debris; organisms found will be added to the sample residue.

+++END-IF+++

1. +++IF determine('Freshwater Benthic', 'Freshwater' , 'Stream characteristics', '') === true+++

## B2 Sampling Method—Physical Habitat Assessment

### B2.1 Method Overview

The condition of the habitat at a biomonitoring station is evaluated using a series of physical parameters. Each of these parameters is numerically scored after visual observation of the stream reach. The numerical scores for all parameters are then summed and the value obtained places the stream within a category ranging from poor to optimal. The assessment is the visual method described by (EPA, 1999).

### B2.2 Equipment List

* Field sensor (for water chemistry and flow velocity)
* Meter stick, meter tape, or range finder
* Digital camera
* Physical Characterization and Habitat Assessment Form (attached)
* Clipboard
* Pencils or waterproof pens
* Upstream/downstream “arrows” or signs for photographing and documenting sampling reaches
* GPS unit

### B2.3 Procedure

1. Perform the habitat assessment on the same reach from which the biological sampling is conducted. Some parameters require an observation of a broader section of the catchment than just the sampling reach.
2. Complete the station identification section of the Physical Characterization Habitat Assessment Form. It is best for the investigators to look closely at the habitat features to make an adequate assessment. If the physical and water quality characterization and habitat assessment are done before biological sampling, care must be taken to avoid disturbing the sampling habitat.
3. Complete the **Physical Characterization Habitat Assessment Form (attached)**, including a sketch of the sampling reach.
4. Complete the **habitat assessment Field Data Form** in a team of two or more biologists, if possible, to come to a consensus on determination of quality. Those parameters to be evaluated on a scale greater than a sampling reach require traversing the stream corridor to the extent deemed necessary to assess the habitat feature. As a general rule of thumb, use two lengths of the sampling reach to assess these parameters.

Habitat assessment data can also be interpreted by summing the habitat parameter scores for an overall assessment value.[[9]](#footnote-10) The categories are (descriptions from the Maine Department of Environmental Protection):

1. **Epifaunal substrate/available cover.** This evaluation includes the relative quantity of natural resources in the stream, such as cobble (riffles), large rocks, fallen trees, logs and branches, undercut banks, available refugia, feeding, or sites for spawning and nursery functions of aquatic macrofauna.

2a. **Embeddedness (low-gradient streams).** The degree that voids between dominant substrates found within riffle run habitats are filled with smaller sized particles is termed embeddedness. As these voids become filled, important microhabitats for benthic dwelling insects and fish are eliminated, and the ecological health and integrity of the area is compromised. In addition to the loss of microhabitat, the substrates ability to entrap coarse particulates such as leaves, and other riparian generated detritus is also reduced, resulting in the loss of important food resources for many locally dwelling organisms.

2b. **Pool substrate characterization (high-gradient streams).** Evaluates the type and condition of bottom substrates found in pools. Firmer sediment types like gravel, sand, and rooted aquatic plants provide support for a more diverse group of organisms than a pool that is dominated by mud, bedrock, and no plants. Additionally, a stream that has a uniform substrate in its pool will not support as many types of organisms as a stream that has a variety of substrate types.

3a. **Velocity/depth regimes (low-gradient streams).** There are basically four types of velocity/depth regimes possible in a river system: deep and slow moving, deep and fast moving, shallow and slow moving, and shallow and fast moving. The more of these velocity/depth regimes that are present in a river or stream, the more varied the habitat and the more amenable to supporting a diverse aquatic community. In larger river systems where only one of these regimes may be commonly present (i.e., deep and slow moving), a different habitat assessment form is used so that the water body is not assessed negatively for naturally occurring conditions.

3b. **Pool variability (high-gradient streams).** This rates the overall mixture of pool types found in streams, according to size and depth. There are four basic types of pools: large-shallow, large-deep, small-shallow, and small-deep. A stream will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community.

4. **Sediment deposition.** Sediment deposition can be caused from increased stream velocities resulting from alteration of the stream channel. Increased stream velocities accelerate the erosion process, increasing suspended materials and bed load sediments (those particles that bounce along the bottom), which are then deposited in lower velocity-areas of the water body. Increasing sediment loads and subsequent deposition into other reaches often results in the covering and encapsulation of coarser streambed materials or the filling of interstitial spaces between the larger substrates that previously provided important habitat for fish and aquatic insects.

5. **Channel flow status.** This parameter represents the degree to which the channel is filled with water. It provides an assessment of the temporal variability of streamflow in the channel and can be related to the suitability of the habitat for inhabitance by fish and aquatic insects. Factors such as hydropower, drinking water diversions, flood control structures, and urban development can precipitate highly varying seasonal and non-seasonal flow regimes, which can reduce the amount of available habitat or alter its characteristics as to be unsuitable for use by the naturally occurring biological community.

6. **Channel alteration.** Channel alteration is an assessment of the degree of diversion from the natural course of the water body by manmade structures and/or activities. This includes rip-rap stream banks, bridge abutments, dredging, concrete channelization, etc. These structures and activities often degrade habitat by increasing stream velocities and decreasing food sources and protective cover. Elimination of streambank vegetation, undercutting of banks, removal of snags, and smothering or elimination of bottom substrates and detritus are all results of channel alteration. Depositional and erosional areas within the river system are often increased or decreased as a result of channel alteration, causing shifts in the structure of the naturally occurring community.

7a. **Riffle frequency (low-gradient streams).** Riffle habitat is considered to be the instream geomorphic feature that provides the most optimal habitat conditions and reflects the balance between erosional and depositional characteristics in the water body. Five to seven stream widths between each recurring riffle area are considered to be optimal.

7b. **Channel sinuosity (high-gradient streams).** Evaluates the meandering or sinuosity of the stream. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides for refugia for benthic invertebrates and fish during storm events.

8. **Bank stability.** Unstable banks, while naturally occurring under some conditions, usually alludes to highly fluctuating flows and the inability of the riparian habitat to recover from frequently occurring hydrologic stresses. Poor bank stability increases turbidity and depositional/erosional areas. It can also elevate instream water temperatures, and cause community shifts from pollutant sensitive aquatic species to pollutant tolerant ones. Poor streamside bank conditions usually coincide with poor instream habitat.

9. **Bank vegetative protection.** Stream-side vegetation is one of the principal factors that protects the streambank from erosional processes, provides shade and protective cover for aquatic life, and provides a significant food source to instream biota. The density and types of vegetation present are indicative of the sensitivity of the water body to potential changes in streamflow and its susceptibility to erosion and sedimentation.

10. **Riparian vegetative zone width.** This habitat quality parameter assesses the width of naturally occurring vegetation between the water body and the area of human land uses in order to determine the riparian zones ability to “buffer” detrimental influxes into the water body. The wider the buffer zone, the greater the ability of the riparian zone to mitigate pollutants. A width of approximately 18 m is considered optimal; additional widths will in most cases not result in additional protection or attenuation of pollutants.

## B3 Sample Handling and Custody

No samples are collected for physical habitat assessment.

## B4 Analytical Methods

No samples are collected for physical habitat assessment.

## B5 Quality Control—Physical Habitat Assessment

Multiple observers (at least two, and ideally three) will perform the habitat assessment at each biomonitoring station. Habitat assessment training will be required to minimize variability in final conclusions. A standardized Habitat Assessment Form will be completed at all biomonitoring stations. Disagreement in habitat parameter scoring will be discussed and resolved before the Habitat Assessment Form can be considered complete.

+++END-IF+++

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

The level of decontamination depends on the sensitivity of the water body to be sampled, as indicated below. Procedures for decontaminating biomonitoring sampling equipment prior to sampling are adapted from Maine Department of Environmental Protection document [DEPLW-0919A-2014](https://www.maine.gov/dep/water/monitoring/biomonitoring/materials/sop_dea_decontamination.pdf), *Protocols for Decontaminating Biomonitoring Sampling Equipment*.

### B6.1 Equipment

* Disinfectant (see notes below)
* For non-absorbent materials (boats, rubber waders, and other “hard-sided” objects), use 2% household bleach solution (2.5 oz bleach per gallon of water).
* For absorbent materials (nets, felt-soled waders, sandals with fabric straps and other “soft-sided” objects), use a 5% quaternary ammonia (Sani-Care Quat-128, etc.) solution (6.5 oz quaternary ammonia per gallon of water).
* Backpack sprayer, garden sprayer, or other suitable applicator
* Scrub brush
* Liquid dish or hand soap (phosphate-free and biodegradable)
* Measuring cup (with cup and ounce increments marked)
* Plastic bucket (to rinse small items)
* 5-gallon plastic container of tap water
* Rubber gloves (including an extra pair)
* Goggles
* Plastic apron (optional)

Notes:

* Always wear gloves and safety goggles when using disinfectant, and avoid contact with exposed skin, clothing, vehicle upholstery, and/or other fabric.
* New bleach solution must be made up daily. New quaternary ammonia solution should be made up every two or three days, or as needed. Old unused solutions must be disposed of down a drain connected to a wastewater treatment system; slowly pour the unused solution down the drain with the tap water running.
* For safety and logistical reasons, only take one type of disinfectant into the field. It is up to the Project Manager to decide which one will be needed based on the types of equipment to be used in the field (absorbent vs. non-absorbent).

### B6.2 Procedure

Level 1 decontamination should always be done. The necessity of decontamination beyond Level 1 is to be determined by the Project Manager, based on the sensitivity of the water body to be sampled.

#### Level 1—Visual Inspection

Applicable to all waters:

1. Visually inspect all equipment having contact with the water (waders, nets, sieve buckets, canoe, boat trailers, etc.) for any plant fragments or other debris. If any plant material or associated mud is found, remove it and either place it in a trashcan or dispose of it on high, dry ground. Do not put it back into the water body or along the shore. All plant fragments must be removed before equipment is transported to another water body.

Allow all equipment to air dry and visually inspect again, repeating procedures if necessary.

#### Level 2 (Done in Addition to Level 1)—Cleaning

Applicable to waters used for aquaculture activities, waters within an Area of Critical Environmental Concern (ACEC) waters designated Statutory Class A or B, or as deemed necessary by the Project Manager:

1. Visually inspect all equipment having contact with the water for any plant fragments or other debris, as outlined for Level 1 visual inspection above.
2. Designate a grassy area or other upland vegetated area, at least 100 ft from open water and remove mud and other debris, by washing with soap and water. Rinse with clean water: either tap water or de-ionized water, as determined by the Project Manager.
3. Allow all equipment to air dry and visually inspect again, repeating procedures if necessary.

#### Level 3 (Done in Addition to Levels 1 and 2)—Cleaning and Disinfection

Applicable to vernal pools, designated salmon rivers, waters designated Statutory Class AA, areas with a known infestation of infectious salmon anemia virus (ISAV), areas with a known infestation of an invasive aquatic plant, areas susceptible to *Didymo* infestation, or as deemed necessary by the Project Manager.

1. Visually inspect all equipment having contact with the water for any plant fragments or other debris, as outlined in Level 1 visual inspection above.
2. Designate a grassy area or other upland vegetated area, at least 100 ft from open water; remove mud and other debris by washing with soap and water and rinsing, as described in Level 2 above.
3. Disinfect by thoroughly spraying all equipment with appropriate disinfectant. Bleach solutions are not recommended for absorbent materials due to ineffective penetration compared to quaternary ammonia solutions.
4. Allow all equipment to air dry and visually inspect again, repeating procedures if necessary.
5. All equipment used to collect water samples (dipper, mixing jugs) must be rinsed three times prior to reuse. Rinse with clean water: either tap water or de-ionized water, as determined by the Project Manager.
6. Sampling devices that are placed into a water body for an extended length of time (e.g., rock bags) will be decontaminated using one of the following methods, as determined by the Project Manager.
7. Air dried for several months (in direct sunlight for part of the time, if possible), or
8. Cleaned with hot soapy water, rinsed with hot tap water, and air dried for several months (in direct sunlight for part of the time, if possible)

## B7 Instrument Calibration and Frequency

**[extract from Freshwater water quality QAPP, Section B7]**

## B8 Inspection/Acceptance of Supplies and Consumables

The Project Manager will be responsible for ensuring correct sample handling by:

* Ensuring availability of all required sampling supplies in the field.
* Properly labeling all sample containers for biological samples in the field.
* Recording all relevant sampling information on the respective field forms and chain-of- custody forms.
* Coordinating the transfer of all samples from the field to laboratories for analysis.

Table B8.1. Critical Field Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| --- | --- | --- |
| Jars for macrofaunal samples | Visually inspected for cracks, breakage, and cleanliness. May be reused. |  |
| 95% ethanol | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment | Visually inspected for obvious defects, damage, and contamination. |  |
| Navigation instruments, digital camera | Functional checks to ensure proper calibration and operating capacity. |  |

## B9 Data Acquisition Requirements (Non-direct Measurements)

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records
* Survey completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (<https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments>).

Secondary data used will be documented according to Section A9 and C2.

## B10 Data Management

### B10.1 Data Custody

Custody of field data will be the responsibility of the Chief Scientist during the field activity. Field data will be recorded electronically or manually on the Field Data Forms.

Laboratory managers will be responsible for custody of data generated by contracted laboratories.

Each team member involved in this project is responsible for the internal custody of their electronic and hard-copy data until they are submitted to the Project Manager. All hand-entered data that are submitted electronically will receive 100% verification prior to submission, and will be entered and checked using double data entry. Formats designed to comply with rules of the WQX web database will be used in the application to constrain data entry. These features will ensure that any entry errors are caught and corrected as the operator keys the data.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater','','') === true+++

# Section B. Marine/Benthic Data Generation and Acquisition

## B1 Sampling Design

The rationale for the sampling design is provided in Section A6. Benthic monitoring is used to determine current benthic community conditions and, with repeated monitoring, long-term trends in sediment quality and benthic communities over time. Sampling sites for this monitoring program are at docks or piers accessible to trained staff and volunteers and suitable for dock-side sample processing without interference from non-participants.

For general health: Sampling sites will be selected for general baseline monitoring, at representative sites around the embayment of interest.

For targeted monitoring: Sampling sites will be selected to assess the impact of a suspected stressor.

## B2 Sampling

All ecological sampling activities performed for benthic monitoring will be conducted following a Massachusetts Division of Marine Fisheries Scientific Collector’s Permit and any local permits that are required. The Project Manager will request the appropriate permits to allow sampling; a copy will be provided to the Chief Scientist prior to the survey.

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

Table B2.1. Processing and Storage of Field Samples taken on Marine Benthic Monitoring Surveys

| Activity | Sediment and Infaunal Survey |
| --- | --- |
| Stations | See Survey Plan |
| Station location and time | Record location, time of station visit, and location of individual samples |
| Weather/sea state/ bottom depth | Record general conditions; record bottom depth to nearest 0.5 m |
| Sampling: Gear | 0.04-m2 Ted Young-modified Van Veen grab sampler |
| Sampling: Measurements | Record penetration depth to nearest 0.5 cm and sediment volume to nearest 0.5 L  Water quality profile: temperature, DO, salinity, pH |
| Sampling: Sediment texture, color, odor | Describe qualitatively |
| Faunal Samples: Processing | Rinse over 500-µm-mesh sieve; fix with 90% ethanol to a final approximate concentration of 70% ethanol |
| Faunal Samples: Storage | Clean, labeled glass or plastic jar; ambient temperature |
| Chemistry Samples: Number | 1 at each station |
| Chemistry Samples: Processing | Use a scoop to collect upper 0–2 cm from the grab, homogenize, and collect ~500 mL subsample for grain size and ~50 mL for TOC. |
| Chemistry Samples: Storage | Clean, labeled, wide-mouth glass jar (500 ml (16 oz) for grain size and 125 ml (4 oz) for TOC); refrigerate grain size, freeze TOC.  Holding time is 28 days for both grain size and TOC. |

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

**Table B2.1. Processing and Storage of Field Samples taken on Marine Benthic Monitoring Surveys**

| Activity | Infaunal group only Survey |
| --- | --- |
| Stations | See Survey Plan |
| Station location and time | Record location, time of station visit, and location of individual samples |
| Weather/sea state/ bottom depth | Record general conditions; record bottom depth to nearest 0.5 m |
| Sampling: Gear | 0.04-m2 Ted Young-modified Van Veen grab sampler |
| Sampling: Measurements | Record penetration depth to nearest 0.5 cm and sediment volume to nearest 0.5 L  Water quality profile: temperature, DO, salinity, pH |
| Sampling: Sediment texture, color, odor | Describe qualitatively |
| Faunal Samples: Processing | Rinse over 500-µm-mesh sieve; fix with 90% ethanol to a final approximate concentration of 70% ethanol |
| Faunal Samples: Storage | Clean, labeled glass or plastic jar; ambient temperature |
| Chemistry Samples: Number | NA |
| Chemistry Samples: Processing | NA |
| Chemistry Samples: Storage | NA |

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === false+++

**Table B2.1. Processing and Storage of Field Samples taken on Marine Benthic Monitoring Surveys**

| Activity | Grain size and/or TOC only Survey |
| --- | --- |
| Stations | See Survey Plan |
| Station location and time | Record location, time of station visit, and location of individual samples |
| Weather/sea state/ bottom depth | Record general conditions; record bottom depth to nearest 0.5 m |
| Sampling: Gear | 0.04-m2 Ted Young-modified Van Veen grab sampler |
| Sampling: Measurements | Record penetration depth to nearest 0.5 cm and sediment volume to nearest 0.5 L  Water quality profile: temperature, DO, salinity, pH |
| Sampling: Sediment texture, color, odor | Describe qualitatively |
| Faunal Samples: Processing | NA |
| Faunal Samples: Storage | NA |
| Chemistry Samples: Number | 1 at each station |
| Chemistry Samples: Processing | Use a scoop to collect upper 0–2 cm from the grab, homogenize, and collect ~500 mL subsample for grain size and ~50 mL for TOC. |
| Chemistry Samples: Storage | Clean, labeled, wide-mouth glass jar (500 ml (16 oz) for grain size and 125 ml (4 oz) for TOC); refrigerate grain size, freeze TOC.  Holding time is 28 days for both grain size and TOC. |

+++END-IF+++

## B4 Analytical Methods

+++IF determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

Macrobiological measures (community measures such as abundance, numbers of species, and diversity) are based on the species-level identifications of the soft-bottom infauna.

Table B4.1. Marine Benthic Survey Sample Analyses, Infauna

| Parameter | Unit of Measurement | Method | Reference |
| --- | --- | --- | --- |
| Infaunal Analysis | Count/species  (# per grab) | ID and Enumeration | Sweeny and Rutecki, 2019 |

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true +++

Sediment geophysical properties, including sediment grain size and total organic carbon, are based on laboratory assessment of soft-bottom grab samples.

Table B4.1 Marine Benthic Survey Sample Analyses, Sediment

| **Parameter** | **Unit of Measurement** | **Method** | **Reference** |
| --- | --- | --- | --- |
| TOC | %C by dry weight | Lloyd Kahn | Kahn, 1988[[10]](#footnote-11) |
| Sediment Grain Size | % dry weight | Folk, 1974[[11]](#footnote-12)  FGDC, 2012[[12]](#footnote-13) | Sweeny and Rutecki, 2019[[13]](#footnote-14) |

+++END-IF+++

## B8 Inspection/Acceptance of Supplies and Consumables

Critical supplies for field activities will be the responsibility of the Project Manager.

If unacceptable supplies or consumables are found, the Project Manager may repair or replace measurement equipment and/or replace defective or inappropriate materials.

+++IF determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

Table B8.1. Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| --- | --- | --- |
| Jars for macrofaunal samples | Visually inspected for cracks, breakage, and cleanliness. May be reused. |  |
| Chemicals and reagents | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment (grabs) | Visually inspected for obvious defects, damage, and contamination. |  |

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true || determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true +++

Table B8.2. Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| --- | --- | --- |
| Sample bottles for sediment chemistry | Visually inspected upon receipt for cracks, breakage, and cleanliness. Must be accompanied by certificate of analysis. |  |
| Chemicals and reagents | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment (grabs) | Visually inspected for obvious defects, damage, and contamination. |  |

+++END-IF+++

## B9 Secondary Data

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (<https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments>)

Secondary data used will be documented in the Secondary Data Form, attached, according to Section A9 and C2.

## B10 Data Management

### B10.1 Sample Analysis

#### Infauna

The contracted laboratory will include the scientific name for each taxon in the macrofaunal abundance data submitted to the Project Manager.

Macrofaunal data will be analyzed for the following community parameters: abundance, Shannon-Wiener diversity index (H'), Pielou's evenness (J'), Margalef’s diversity index, Simpson, and/or Total Taxonomic Distinctness (TTD).

Shannon-Wiener diversity index (H') characterizes the species diversity in a community and is calculated following

H’ =

where *p*i is the proportion of individuals belonging to the *i*th species in the sample.

Pielou's evenness is calculated by

J’ = H’/H’max

where H’ is derived from the Shannon-Wiener diversity index and H’max is the maximum possible value of H’ (if every species was equally likely), calculated by H’ = ln *S*. *S* is species richness, the total number of species in the sample. J’ ranges between 0 and 1, the lower J’ is the less evenness is a community between the species.

Margalef’s diversity index (DMg ) is calculated by

DMg = (*S* - 1)/ln N

where *S* is species richness and N is the total number of individuals in the sample.

TTD can be calculated[[14]](#footnote-15) which can be used to document change in taxonomic distinctness with increased stress.[[15]](#footnote-16) Distinctness is different from species diversity in that it describes the phylogenetic distance between observed species, as well as the diversity of function.

The results of all statistical analyses will be combined and tabulated into an Excel spreadsheet for delivery to the Project Manager.

#### Sediment

The contracted laboratory will include sediment grain size and percentage of total organic carbon in the sediment data submitted to the Project Manager. After data verification sediment samples can be disposed of following internal laboratory protocols. Sediment samples with known toxins (e.g., PCBs, dioxin, and PAHs) will be disposed of properly following local, state, and federal laws.

### B10.2 Data Custody

The Field Coordinator will be responsible for custody of field data during the field activity. Field data will be recorded electronically or manually on the Field Data Forms.

Laboratory managers will be responsible for custody of data generated by contracted laboratories (see Section B10.3).

Each team member involved in this project is responsible for the internal custody of their electronic and hard-copy data until they are submitted to the Project Manager. All hand-entered data that are submitted electronically will receive 100% verification prior to submission.

### B10.3 Laboratory Data and Data Reduction

All data generated by contracted laboratories will be either electronically transferred from the instrument or manually read from the instrument display (optical field of a microscope or video monitor) and entered directly into an electronic format or entered into laboratory data forms and then manually entered into an electronic format. All manually entered data will receive 100% verification or will be entered and checked using double data entry.

Data reduction is the process of converting raw numbers (e.g., numbers of organisms per replicate) into data that can be displayed graphically, summarized in tables, or compared statistically for differences between mean values for sampling stations or times. Macrofauna data analysis discussed below require that some data be derived from the raw numbers for the synthesis report. All data reduction will be performed electronically, either by the instrument software or in a spreadsheet, and will be validated according to procedures described in Section D2.

The format for final data submission is described in Sections A9 and C2.

### B10.3 Dataset Structure

Electronic data deliverables will be prepared by the contracted laboratory in a structure and format suitable for upload.

### B10.4 Project Database Codes

Standardized codes and qualifiers will be used to ensure consistency over time.

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

## B2 Marine Benthic Sample Collection, Processing, and Storage

Processing and storage requirements for all samples collected for the benthic monitoring tasks are described in the following sections.

### B2.1 Soft-Bottom Grab Sample Collection

A 0.04 m2 Ted Young-modified Van Veen grab sampler will be used to collect bottom sediment samples. At each station, three grab samples will be collected for infaunal analysis (two to be sorted at the lab and one archived) and one grab sample will be collected for total organic carbon and grain size analysis.

Supply list:

* Young-modified Van Veen grab with grab stand or frame if needed
* Weights and pads for grab
* Nitrile gloves
* Plastic tub or bucket
* 0.500 mm screening bucket or stainless steel sieve
* Sieve box or bucket
* Electrical tape
* Forceps (fine-tipped)
* Funnel (wide-mouth)
* 95% ethanol
* Ruler (cm)
* Squirt bottle (ambient water)
* Stainless steel or glass mixing pot or bowl with lid
* Stainless steel or Teflon spoons (15”), scoops, or spatula
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (500 mL) with screw-cap lids
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (125 mL) with screw-cap lids
* Scrub brush
* Cooler with wet ice (for grain size samples)
* Cooler with dry ice (for total organic carbon samples)

The following items will also be needed for recording measurements:

* Survey log form
* Sample collection form
* Pencils
* Waterproof paper for internal sample jar labels
* Fine-tipped indelible markers (for labels)
* Write-on colored tape or pre-printed write-on labels
* Clear tape strips

Prior to sample collection (before the sample jar gets wet), an External Sample Label (including station location, replicate number, and date) will be taped to the outside of jars for infaunal samples. The external label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. An internal label made of waterproof paper will be filled in onsite with a pencil. Infaunal samples will be preserved in the field using 95% ethanol.

Once the survey crew is at the sampling site, and coordinates have been verified, the sediment grab will be deployed.

1. Attach the sampler to the end of the winch cable with a shackle and tighten the pin (or secure the pin with a cable tie).
   1. Set the grab according to the manufacturer’s instructions and disengage any safety device designed to lock the sampler open.
   2. Lower the grab sampler through the water column no faster than about 1 m/second. This minimizes the effects of wave disturbance on surficial sediments.
   3. Allow a moment for the sampler to settle into the substrate and then allow slack on the cable (letting the cable go slack serves to release the jaws of the sampler so they will close as the sampler is retrieved).
   4. Retrieve the sampler and lower it into its cradle or a plastic tub on the dock or pier. Open the top and determine whether the sampling is successful or not (see Figure A7.1).
2. A successful grab is one having relatively level, intact sediment over the entire area of the grab, and a sediment depth at the center of at least 7 cm for the benthic macroinvertebrate grab.
   1. Grabs containing no sediment, partially filled grabs, or grabs with shelly/rocky substrates or grossly slumped surfaces are unacceptable.
   2. Grabs completely filled to the top, where the sediment is in direct contact with the hinged top, are also unacceptable.
   3. It may take several attempts using different amounts of weight to obtain the first acceptable sample. More weight will result in a deeper bite of the grab. In very soft mud, pads may be needed to prevent the sampler from sinking into the mud. If pads are used, the rate of descent near the bottom should be slowed even further to reduce the wave.
   4. If, after several attempts, only grabs less than 7 cm deep can be obtained, use the next successful grab regardless of the depth of sediment at the center of the grab.
3. Use the comments section on the sample collectionform to describe sampling efforts and accurately record the depth of the sediment captured by the grab.
   1. Carefully drain overlying water from the grab. If the grab is used for benthic infaunal analysis, the water must be drained into the container that will receive the sediment to ensure that no organisms are lost.
   2. Enter notes on the condition of the sample (smell, substrate, presence of organisms on the surface, etc.) on the sample collection **f**orm.
4. Process the grab sample for either benthic community analysis or sediment testing as described below.
5. Take precautions during the deployment and retrieval of the grab sampler to prevent contamination of samples between stations. Sampling for grain size, total organic carbon, and infauna determinations requires that the grab and associated sampling equipment be washed and rinsed with screened ambient seawater until free of all sediment.

The penetration depth, sediment volume, and sediment texture will be visually estimated for acceptable samples. These data will be recorded in the field log. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

#### Penetration Depth

The penetration depth of the sample will then be measured using a plastic ruler (marked in mm) pushed into the sediment. Any sediment adhering to the surface of the ruler will be rinsed back into the grab for processing with the remainder of the sample.

#### Sediment Volume

The volume of the grab will be estimated by comparing the measured penetration depth according to Table B2.1.

Table B2.1. Values Used to Convert Grab Penetration Depth to Sediment Volume

|  |  |
| --- | --- |
| Grab Penetration Depth (cm)1 | Sediment Volume (L) 0.04-m2 Grab |
| 4.1-5.0 | 1.4 |
| 5.1-6.0 | 1.8 |
| 6.1-7.0 | 2.1 |
| 7.1-8.0 | 2.4 |
| 8.1-9.0 | 2.7 |
| 9.1-10.0 | 3.0 |
| 10.1-11.0 |  |
| 11.1-12.0 |  |
| 12.1-13.0 |  |
| 13.1-14.0 |  |
| 14.1-15.0 |  |

1Over penetration is > 9.5 cm for 0.04-m2 grab.

#### Sediment Texture

The sediment should be characterized as being coarse sand, fine sand, silt, clay, gravel, mud, or of a mixed type. The presence of shell hash should also be recorded.

#### Sample Processing

##### Infaunal Samples

If the grab sample is acceptable, the sample will be processed. For the macrofauna samples only, photographs of the grab will be taken before and after the sample has been sieved. Sample labels (station location, replicate number, and date) will be included in each photograph to identify the sample. The first photograph of the macrofaunal sample will be taken prior to measuring penetration depth. Once the photo is taken, large macrofauna or epifauna will be removed and not counted in the sample total (e.g., sand dollars, anemones, shrimp, sea cucumbers, snails, clams, mussels, hermit crabs). After penetration depth measurements are taken, the grab will be placed over a bucket, the jaws opened, and the sample emptied into a bucket. Screened ambient seawater will be used to gently wash the sample into the bucket. Once thoroughly washed, the grab will be redeployed to collect a total of three samples for infaunal analysis. The screening bucket or sieve will be washed between samples using copious amounts of forceful water and a stiff brush.

Grab samples for infaunal analyses will be rinsed with clean seawater through a 500 µm mesh sieve. Once the sample has been sieved, a second photograph of the sample will be taken. The portion retained on the screens will be transferred to the labeled jars and fixed with 95% ethanol (final ethanol concentration approximately 70%). Fine forceps can be used, if necessary, to transfer infauna to the sample jar. Each sample jar will be filled no more than half full of material and filled to within 1 cm of the top with seawater to prevent infauna from sticking to the sample jar top, then gently rotated to distribute the ethanol evenly throughout the sample. Sample jars will be labeled with external and internal labels. The lids on the sample jars will be taped and the jars inserted individually into large zip-locked or tied plastic bags lined with absorbent padding.

The infaunal samples (stored in sturdy coolers) will be delivered to the contracted laboratory within 48 hours.

##### Sediment Sample Processing - Grain Size and Total Organic Carbon Samples

A separate grab sample will be collected for sediment analysis. When a sample is collected that meets the acceptability criteria, the water overlying the sample will be siphoned from the grab and the surface sediment (top 0 to 2 cm) will be collected with a scoop and transferred to a clean (rinsed with clean seawater) stainless steel or glass bowl. The sediment will be thoroughly homogenized, then two subsamples will be collected:

* Subsamples of approximately 500 mL for grain size analysis in 500 mL (16 oz) wide-mouth sample jars. These samples will be labeled and kept on ice at 1 to 4C for delivery to the laboratory.
* Subsamples of approximately 50 mL subsamples for total organic carbon analysis in 125 mL (4 oz) wide-mouth sample jars. These samples will be labeled and kept on dry ice (frozen) for delivery to the laboratory.

These samples will be delivered to the contracted laboratory for analysis within 24 hours of survey completion.

The maximum holding time in the laboratory for grain size samples will be 28 days with refrigeration and samples for TOC analysis will be 28 days frozen. These time frames are consistent with several standard EPA Methods and ensures that samples are analyzed in a timely manner to prevent or minimize analyte degradation and interferences.

## B3 Sample Handling and Custody

### B3.1 Sample Handling

Section B2 describes handling of samples while in the field, including storage requirements.

Following the soft-bottom benthic survey, the infaunal samples (stored in sturdy coolers) will be delivered to the contracted laboratory according to a pre-arranged schedule for sample dropoff. The samples, while still in approximately 70% ethanol, may be shipped by ground (or two-day express delivery if necessary) for delivery to the contracted laboratory. The lids on the sample jars will be taped and the jars inserted individually into large zip-locked or tied plastic bags lined with absorbent padding. At the laboratory, one sample from each infaunal station will be randomly selected to archive (see Section B3.2) and the other two will be processed. The organisms will be picked from the samples and sorted into major taxonomic groups.

The sediment chemistry samples collected during the benthic survey must be kept cold (sediment grain size samples) or frozen (total organic carbon samples). After the survey is completed, a survey crew member will deliver the sediment chemistry samples to the contracted laboratory according to a pre-arranged schedule for sample dropoff. If circumstances dictate that the samples must be shipped to the laboratory, they will be shipped by overnight express. In that case, the samples that were frozen after collection will be placed on dry ice with protective layers of foam or bubble wrap to ensure that they remain intact and frozen during shipment.

### B3.2 Sample Custody

#### Sample Tracking

Sample custody will be tracked through external and internal sample labels, sample collection forms, and chain of custody forms.

Sediment samples collected under this QAPP will be processed by a contracted laboratory. The contracted laboratory will provide the sample containers and sample labels. Sample labels will contain or have spaces for following information: station location, survey type, analysis, preservative, date/time collected, and collector’s name. The Chief Scientist is responsible for verifying that information on the sample labels matches the information on the chain of custody prior to delivering the samples to the contracted laboratory.

The survey crew will fill out the sample collection form at each station. The form includes headers for entering pertinent information about each station, such as arrival time, bottom depth, and weather observations. The form sheets also contain spaces for specific grab data, such as penetration depth and general descriptions. The sheets will remain in the survey logbook and will be kept in the project files. During field collection, chain of custody forms also will be completed. The chain of custody forms will include the unique information from the corresponding label on the sample container, ensuring the tracking of sample location and status.

#### Sample Custody

Samples will be in the custody of the survey Chief Scientist or a crew member from collection until they are transferred to the contracted laboratory. Transfer of samples will be documented on the custody forms. All samples will be distributed to the appropriate laboratory personnel by hand or by a shipping service. A copy of the chain of custody form will be retained by the field sample custodian in the field log. The original will accompany the samples to the laboratory for subsequent sample transfer. When samples arrive at the laboratory, custody will be relinquished to the laboratory staff. The laboratory staff will verify that the custody seals on the cooler are intact. The laboratory staff will then examine the samples, verify that sample-specific information recorded on the chain of custody form is accurate and that the sample integrity is uncompromised, log the samples into their laboratory tracking system, and complete and sign the chain of custody form so that transfer of custody of the samples is complete. Any discrepancies between sample labels and transmittal forms, the condition of the samples upon receipt, and any unusual events or deviations from the QAPP will be documented in detail on the chain of custody form, and the Project Manager notified. Copies of completed chain of custody forms will be delivered (scanned and emailed or faxed) to the Project Manager within 24 hours of receipt.

## B4 Analytical Methods

Sediment geophysical properties, including sediment grain size and total organic carbon, are based on laboratory assessment of soft-bottom grab samples.

Table B4.1 Marine Benthic Survey Sample Analyses, Sediment

| **Parameter** | **Unit of Measurement** | **Method** | **Reference** |
| --- | --- | --- | --- |
| TOC | %C by dry weight | Lloyd Kahn | Kahn, 1988[[16]](#footnote-17) |
| Sediment Grain Size | % dry weight | Folk, 1974[[17]](#footnote-18)  FGDC, 2012[[18]](#footnote-19) | Sweeny and Rutecki, 2019[[19]](#footnote-20) |

## B5 Soft-Bottom Grab Sampling Quality Control

All samples will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. A single grab sample, collected by this grab sampler, will provide adequate quantities of sediment for grain size and total organic carbon analysis. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment.
* Control of penetration by adding or removing weights to the frame and adjusting descent rate.
* Slow recovery until the grab is free of the bottom.
* Inspection for signs of leakage.
* Securing the grab on the dock or pier.

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* Sampler is not overfilled with sediment, the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors.
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable according to the following standards:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside of that grid.
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

### B5.1 Sampling Quality Control for Benthic Infauna

#### Accuracy, Precision, and Representativeness

There will be no subsampling. Consequently, the accuracy, precision, and representativeness of the sampling will depend upon the factors discussed above under Section A7.

#### Comparability

Procedures for washing, sieving, and preserving the samples will be consistent with methods described in Section B2. Samples will be collected only by trained staff, volunteers under the supervision of a staff person, or volunteers with experience in the collection of benthic infaunal samples.

#### Completeness

All required samples will be collected at all of the stations specified in the sampling design. The entire sample will be sieved, and all material retained on the 500 µm mesh screen will be fixed for analysis.

### B5.2 Sampling Quality Control for Sediment

#### Accuracy, Precision, and Representativeness

These qualities will be ensured by the sampling plan and by ensuring that samples are well homogenized and subsampled and preserved following methods detailed in Section B2.

#### Comparability

Procedures for collecting and preserving the samples will be consistent with methods described in Section B2. Procedures for sampling and subsampling are comparable to those used in other investigations in Massachusetts coastal waters.

#### Completeness

All required samples will be collected at all of the stations specified in the monitoring program sampling design.

### B5.3 Benthic Analysis Laboratory Quality Control

#### Benthic Infauna

Details on infaunal sample analysis methods to be undertaken by taxonomists are provided in the selected laboratory’s QAPP (attached).

##### Accuracy

Benthic macrofauna will be identified by experienced taxonomists at a contracted laboratory. In cases where different taxonomists identify replicates from the same station, discrepancies in species identifications will be recognized during data entry and reviewed. Taxonomic discrepancies will be addressed by communication among the taxonomists. In the case of questions about organisms in specific taxonomic groups, specimens may be sent to recognized experts for a second opinion on the identification. Standard taxonomic references will be used, and selected specimens of newly found species will be retained as part of the reference collection.

##### Precision

Sorting technicians will remove all organisms from the samples and separate them into major taxonomic groups. All residual material will be labeled and stored for QC analysis. Samples will be divided into batches of approximately 10. Approximately 10% of the samples from each batch will then be randomly chosen for an independent QC check. If more than 5% of the total organisms in the QC sample have been missed, all remaining samples from that batch will be re-sorted.

##### Representativeness

Because all of the sample will be analyzed, representativeness will be determined by sampling factors.

##### Completeness

Since one sample from each station will be archived, the loss of one sample will still permit data to be obtained from the archived sample for that station. One hundred percent completeness is expected.

##### Comparability

Methods of analysis will be comparable to those used in other investigations conducted in Massachusetts coastal waters. Comparability of the identifications will be ensured through use of standard taxonomic references. Taxonomists will be familiar with fauna from Massachusetts waters and those of the surrounding regions. A reference collection will be maintained and, if new species are identified, expanded. Any new species that have not been reported in previous studies conducted in the Massachusetts coastal waters will be checked against similar taxa in the reference collection and carefully verified with recognized experts.

#### Sediment

##### Accuracy

Sediment samples collected will be analyzed for sediment grain size and TOC by a contracted laboratory. No field-collected QC samples, including field duplicates, or equipment and field blanks for sediment chemistry are required.

Details on sediment sample analysis methods are provided in the selected laboratory’s QAPP (attached). The laboratory will follow all QC/QA procedures for the analytical method being followed.

##### Representativeness

Because all of the sample will be analyzed, representativeness will be determined by sampling factors.

##### Completeness

Adequate sediment will be collected for the analytical laboratories to perform the required analyses.

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

No analytical laboratory instruments are covered by this QAPP.

## B7 Instruments

No analytical laboratory instruments are covered by this QAPP.

## B8 Inspection/Acceptance of Supplies and Consumables

Critical supplies for field activities will be the responsibility of the Project Manager.

If unacceptable supplies or consumables are found, the Project Manager may repair or replace measurement equipment and/or replace defective or inappropriate materials.

Table B8.1. Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

|  |  |  |
| --- | --- | --- |
| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| Jars for macrofaunal samples | Visually inspected for cracks, breakage, and cleanliness. May be reused. |  |
| Sample bottles for sediment chemistry | Visually inspected upon receipt for cracks, breakage, and cleanliness. Must be accompanied by certificate of analysis. |  |
| Chemicals and reagents | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment (grabs) | Visually inspected for obvious defects, damage, and contamination. |  |

Table B8.2. Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies

|  |  |  |
| --- | --- | --- |
| Critical Supplies and Consumables | Inspection Requirements  and Acceptance Criteria | Responsible Individual |
| Sample bottles for sediment chemistry | Visually inspected upon receipt for cracks, breakage, and cleanliness. Must be accompanied by certificate of analysis. |  |
| Chemicals and reagents | Visually inspected for proper labeling, expiration dates, appropriate grade. |  |
| Sampling equipment (grabs) | Visually inspected for obvious defects, damage, and contamination. |  |

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other studies water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records.
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments).

Secondary data used will be documented in the Secondary Data Form, attached, according to Sections A9 and C2.

## B10 Data Management

### B10.1 Macrofaunal Analysis

The contracted laboratory will include the scientific name for each taxon in the macrofaunal abundance data submitted to the Project Manager.

Macrofaunal data will be analyzed for the following community parameters: abundance, Shannon-Wiener diversity index (H'), Pielou's evenness (J'), Margalef’s diversity index (DMg), Simpson, and/or total taxonomic distinctness.

Shannon-Wiener diversity index characterizes the species diversity in a community and is calculated as:

H’ =

where *p*i is the proportion of individuals belonging to the *i*th species in the sample.

Pielou's evenness is calculated by

J’ = H’/H’max

where where H’ is derived from the Shannon-Wiener diversity index and H’max is the maximum possible value of H’ (if every species were equally likely), calculated by H’ = ln *S*. *S* is species richness, the total number of species in the sample. J’ ranges between 0 and 1, with lower numbers indicating less evenness between the species.

Margalef’s diversity index (DMg) is calculated by

DMg = (*S* - 1)/ln N

where *S* is species richness and N is the total number of individuals in the sample.

Total taxonomic distinctness can be calculated[[20]](#footnote-21) which can be used to document change in taxonomic distinctness with increased stress.[[21]](#footnote-22) Distinctness is different from species diversity in that it describes the phylogenetic distance between observed species, as well as the diversity of function.

The results of all statistical analyses will be combined and tabulated into an Excel spreadsheet for delivery to the Project Manager.

### B10.2 Sediment Physiochemical Analysis

The contracted laboratory will include sediment grain size and percentage of total organic carbon in the sediment data submitted to the Project Manager. After data verification, sediment samples can be disposed of following internal laboratory protocols. Sediment samples with known toxins (e.g., PCBs, dioxin, and PAHs) will be disposed of properly following local, state, and federal laws.

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === true+++

## B2 Benthic Sample Collection, Processing, and Storage *– if infaunal group selected*

Processing and storage requirements for all samples collected for the benthic monitoring tasks are described in the following sections.

### B2.1 Soft-Bottom Grab Sample Collection

A 0.04 m2 Ted Young-modified Van Veen grab sampler will be used to collect bottom sediment samples. At each station, three grab samples will be collected for infaunal analysis (two to be sorted at the lab and one archived).

Supply list:

* Young-modified Van Veen grab with grab stand or frame if needed
* Weights and pads for grab
* Nitrile gloves
* Plastic tub or bucket
* 0.500 mm screening bucket or stainless steel sieve
* Sieve box or bucket
* Electrical tape
* Forceps (fine-tipped)
* Funnel (wide-mouth)
* 90% ethanol
* Ruler (cm)
* Squirt bottle (ambient water)
* Stainless steel mixing pot or bowl with lid
* Stainless steel or Teflon spoons (15”), scoops, or spatula
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (500 mL) with lids\*
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (125 mL) with lids\*
* Plastic bags (e.g., Whirl Pak)
* Scrub brush
* Cooler with wet ice

The following items will also be needed for recording measurements:

* Sampling log
* Sample collection form
* Pencils
* Waterproof paper for internal sample jar labels
* Fine-tipped indelible markers (for labels)
* Write-on colored tape or pre-printed write-on labels
* Clear tape strips

Prior to sample collection (before the sample jar gets wet), an external label (including station location, replicate number, and date) will be taped to the outside of jars for infaunal samples. The external label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Labels made of waterproof paper will be filled in onsite with a pencil. Sample jars will be glass or Nalgene (or other sturdy plastic) jars with screw-capped lids. Samples will be preserved in the field using 90% ethanol (final ethanol concentration approximately 70%).

Once the sampler is on station and coordinates have been verified, the sediment grab will be deployed.

1. Attach the sampler to the end of the winch cable with a shackle and tighten the pin (or secure the pin with a cable tie).
2. Set the grab according to the manufacturer’s instructions and disengage any safety device designed to lock the sampler open.
3. Lower the grab sampler through the water column such that travel through the last 5 m is no faster than about 1 m/second. This minimizes the effects of bow wave disturbance on surficial sediments.
4. Allow a moment for the sampler to settle into the substrate and then allow slack on the cable (letting the cable go slack serves to release the jaws of the sampler so they will close as the sampler is retrieved).
5. Retrieve the sampler and lower it into its cradle or a plastic tub on board. Open the top and determine whether the sampling is successful or not (see Figure A7.1).
   1. A successful grab is one having relatively level, intact sediment over the entire area of the grab, and a sediment depth at the center of at least 7 cm for the benthic macroinvertebrate grab.
   2. Grabs containing no sediment, partially filled grabs, or grabs with shelly/rocky substrates or grossly slumped surfaces are unacceptable.
   3. Grabs completely filled to the top, where the sediment is in direct contact with the hinged top, are also unacceptable.
   4. It may take several attempts using different amounts of weight to obtain the first acceptable sample. More weight will result in a deeper bite of the grab. In very soft mud, pads may be needed to prevent the sampler from sinking into the mud. If pads are used, the rate of descent near the bottom should be slowed even further to reduce the bow wave.
6. If, after several attempts, only grabs less than 7 cm deep can be obtained, use the next successful grab regardless of the depth of sediment at the center of the grab.
   1. Use the comments section on the sample collectionform to describe your efforts and be sure to accurately record the depth of the sediment captured by the grab.
   2. Carefully drain overlying water from the grab into the container that will receive the sediment to ensure that no organisms are lost.
   3. Enter notes on the condition of the sample (smell, substrate, presence of organisms on the surface, etc.) in the “Sediment Characteristics” section of the sample collectionform.
7. Process the grab sample for either benthic community analysis or sediment testing as described below.
8. Take precautions during the deployment and retrieval of the grab sampler to prevent contamination of samples between stations. Sampling for infauna determinations requires that the grab and associated sampling equipment be washed and rinsed with screened ambient seawater until free of all sediment.

The penetration depth, sediment volume, and sediment texture will be visually estimated for acceptable samples. These data will be recorded in the field log. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

#### Penetration Depth

The penetration depth of the sample will then be measured using a plastic ruler (marked in mm) pushed into the sediment. Any sediment adhering to the surface of the ruler will be rinsed back into the grab for processing with the remainder of the sample.

#### Sediment Volume

The volume of the grab will be estimated by comparing the measured penetration depth according to Table B2.1.

Table B2.1. Values Used to Convert Grab Penetration Depth to Sediment Volume

| Grab Penetration Depth (cm)1 | Sediment Volume (L) 0.04-m2 Grab |
| --- | --- |
| 4.1-5.0 | 1.4 |
| 5.1-6.0 | 1.8 |
| 6.1-7.0 | 2.1 |
| 7.1-8.0 | 2.4 |
| 8.1-9.0 | 2.7 |
| 9.1-10.0 | 3.0 |
| 10.1-11.0 |  |
| 11.1-12.0 |  |
| 12.1-13.0 |  |
| 13.1-14.0 |  |
| 14.1-15.0 |  |

1Over penetration is > 9.5 cm for 0.04-m2 grab.

#### Sediment Texture

The sediment should be characterized as being coarse sand, fine sand, silt, clay, gravel, mud, or of a mixed type. The presence of shell hash should also be recorded.

#### Sample Processing

If the grab sample is acceptable, the sample will be processed. For the macrofauna samples only, photographs of the grab will be taken before and after the sample has been sieved. Sample labels (station location, replicate number, and date) will be included in each photograph to identify the sample. The first photograph of the macrofaunal sample will be taken before measuring penetration depth. Once the photo is taken, large macrofauna or epifauna will be removed and not counted in the sample total (e.g., sand dollars, anemones, shrimp, sea cucumbers, snails, clams, mussels, hermit crabs). After penetration depth measurements are taken, the grab will be placed over a bucket, the jaws opened, and the sample emptied into a bucket. Screened ambient seawater will be used to gently wash the sample into the bucket. Once thoroughly washed, the grab will be redeployed until the required numbers of acceptable samples have been obtained for infaunal analysis. The screening bucket or sieve will be washed between samples using copious amounts of forceful water and a stiff brush.

Grab samples for infaunal analyses will be rinsed with clean seawater through a 500 µm mesh sieve. Once the sample has been sieved, a second photograph of the sample will be taken. The portion retained on the screens will be transferred to the labeled jars and fixed with 95% ethanol (final ethanol concentration approximately 70%). Fine forceps can be used, if necessary, to transfer infauna to the sample jar. Each sample jar will be filled no more than half full of material and filled to within 1 cm of the top with seawater to prevent infauna from sticking to the sample jar top. The jar will be gently turned around on its side to distribute the ethanol evenly throughout the sample. The technician sieving each sample will be identified by his or her initials in the survey log. Sieves will be washed between samples. Sample jars will be labeled with external and internal labels. The lids on the sample jars will be taped and the jars inserted individually into large zip-locked or tied plastic bags lined with absorbent padding (Section B3.1).

### B2.2 Sample Storage

All benthic infaunal samples must be handled gently during the sieving process and fixed with 90% ethanol as quickly as possible (final ethanol concentration approximately 70%) to prevent deterioration of the fauna; all sample jars must be labeled accurately. Following each benthic survey, the infaunal samples will be stored in sturdy coolers and delivered to the contracted laboratory within 48 hours.

## B3 Sample Handling and Custody

### B3.1 Sample Handling

Handling of samples while in the field, including storage requirements, is described in Section B2 (see Table 5) above.

Following the soft-bottom benthic survey, the infaunal samples (stored in sturdy coolers) will be delivered by a survey crew member to the contracted laboratory. A crew member will contact laboratory staff to arrange a time for sample dropoff. This will allow laboratory staff to be prepared for sample receipt. The samples, while still in approximately 70% ethanol, can be shipped by ground (or two-day express delivery if necessary) for delivery to the contracted laboratory. The lids on the sample jars will be taped and the jars inserted individually into large zip-locked or tied plastic bags lined with absorbent padding. At the laboratory, one sample from each infaunal station will be randomly selected to archive (see Section B3.2) and the other two will be processed.

### B3.2 Sample Custody

#### Sample Tracking

Sample custody will be tracked through external and internal sample labels, sample collection forms, and chain of custody forms (samples attached).

Sample labels for macrofaunal samples will be affixed to the sample containers in the field. The sampling station, sample type, replicate number, date, and time will be entered manually onto the label. One additional label will be prepared on waterproof paper and inserted inside the sample container. If multiple sample containers are needed for a single infaunal replicate, the sample information will be manually entered on blank labels, and the containers will be numbered (e.g., “1 of 2,” “2 of 2”). Sampling station and replicate number will be printed on the chain of custody forms.

#### Sample Custody

Infauna samples will be in the custody of the survey Chief Scientist or a crew member from collection until they are transferred to the contracted laboratory. Chain of custody forms will accompany the samples. One complete (copied) set of the infauna chain of custody forms will be included in each shipping container and the original chain of custody forms will be returned to Project Manager after the samples have been logged in at the contracted laboratory. The signed original custody forms will be retained in the project files. Sample processing will occur in the contracted laboratory. After the samples are processed, the laboratory will store the appropriate samples and specimens for the specific length of time for re-identification QC, voucher, or unforeseen circumstances.

Transfer of benthic infaunal samples will be documented on the custody forms. All samples will be distributed to the appropriate laboratory personnel by hand or by a shipping service. A copy of the chain of custody form will be retained by the field sample custodian in the field log. The original will accompany the samples to the laboratory for subsequent sample transfer. When samples arrive at the laboratory, custody will be relinquished to the laboratory staff. The laboratory staff will verify that the custody seals on the cooler are intact. The laboratory staff will then examine the samples, verify that sample-specific information recorded on the chain of custody form is accurate and that the sample integrity is uncompromised, log the samples into their laboratory tracking system, and complete and sign the chain of custody form so that transfer of custody of the samples is complete. Any discrepancies between sample labels and transmittal forms, the condition of the samples upon receipt, and any unusual events or deviations from the QAPP will be documented in detail on the chain of custody form, and the Project Manager notified. Copies of completed custody forms will be delivered (scanned and emailed or faxed) to the Project Manager within 24 hours of receipt.

#### Sample Archival Policies

One randomly selected sample from each soft-bottom infaunal station will be archived, and the other two will be processed. Archived soft-bottom infaunal samples will be rinsed with fresh water over 500 µm mesh screens and transferred to reagent alcohol for storage at the laboratory.

Macrofauna samples (both archived and processed samples) will be held until the Project Manager accepts the synthesis report. If subsequent surveys will be conducted within six years, reference collection specimens will be retained by the contracted laboratory. Reference collection specimens will be clearly identified, labeled with the project name and unique identification number, and stored under appropriate conditions for the length of the storage period.

## B4 Soft-Bottom Infaunal Analysis

The contracted taxonomist will carry out identification and enumeration of infaunal species according to the laboratory QAPP which is attached.

## B5 Soft-Bottom Grab Sampling Quality Control

All sediment samples to be used for faunal analyses will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment
* Control of penetration by adding or removing weights to the frame and adjusting descent rate
* Slow recovery until the grab is free of the bottom
* Inspection for signs of leakage
* Securing the grab on the dock or pier

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* The sampler is not overfilled with sediment, the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable according to the follow standards:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside that grid.
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

### B5.1 Soft-Bottom Grab Sampling Quality Control

#### Accuracy, Precision, and Representativeness

There will be no subsampling. Consequently, the accuracy, precision, and representativeness of the sampling will depend upon the factors discussed above under Section A7.1.3.

#### Comparability

Procedures for washing, sieving, and preserving the samples will be consistent with methods described in Section B2. Samples will be collected only by trained staff under the supervision of a Chief Scientist with experience in the collection of benthic infaunal samples.

#### Completeness

All required samples will be collected at all of the stations specified in the project sampling plan. The entire sample will be sieved and all material retained on the 500 µm mesh screen will be fixed for analysis.

All sediment samples to be used for faunal analyses will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment
* Control of penetration by adding or removing weights to the frame and adjusting descent rate
* Slow recovery until the grab is free of the bottom
* Inspection for signs of leakage
* Securing the grab on the dock or pier

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* The sampler is not overfilled with sediment, the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable according to the follow standards:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside that grid.
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

### B5.2 Benthic Infauna Analysis Laboratory Quality Control

Details on infaunal sample analysis methods to be undertaken by taxonomists are provided in the laboratory’s QAPP (attached).

#### Accuracy

Benthic macrofauna will be identified by experienced taxonomists at a contracted laboratory. In cases where different taxonomists identify replicates from the same station, discrepancies in species identifications will be recognized during data entry and reviewed. Taxonomic discrepancies will be addressed by communication among the taxonomists. In the case of questions about organisms in specific taxonomic groups, specimens may be sent to recognized experts for a second opinion on the identification. Standard taxonomic references will be used, and selected specimens of newly found species will be retained as part of the reference collection.

#### Precision

Sorting technicians will remove all organisms from the samples and separate them into major taxonomic groups. All residual material will be labeled and stored for QC analysis. Samples will be divided into batches of approximately 10. Approximately 10% of the samples from each batch will then be randomly chosen for an independent QC check. If more than 5% of the total organisms in the QC sample have been missed, all remaining samples from that batch will be re-sorted.

#### Representativeness

Because all of the sample will be analyzed, representativeness will be determined by sampling factors.

#### Completeness

Since one sample from each station will be archived, the loss of one sample will still permit data to be obtained from the archived sample for that station. One hundred percent completeness is expected.

#### Comparability

Methods of analysis will be comparable to those used in other investigations conducted in Massachusetts coastal waters. Comparability of the identifications will be ensured through the use of standard taxonomic references and by comparison of specimens in the MEP Benthic Monitoring Reference Collection. Taxonomists will be familiar with fauna from Massachusetts waters and those of the surrounding regions. The reference collection will be maintained and, if new species are identified, expanded. Any new species that have not been reported in previous MEP Benthic Monitoring surveys or other studies conducted in the Massachusetts coastal waters will be checked against similar taxa in the reference collection and carefully verified with recognized experts.

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Records

No analytical laboratory instruments are covered by this QAPP.

## B7 Instruments

No analytical laboratory instruments are covered by this QAPP.

Insert Section B8 from page 82

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other studies water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records.
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments).

Secondary data used will be documented in the Secondary Data Form, attached, according to Sections A9 and C2.

## B10 Data Management

### B10.1 Sample Analysis

The contracted laboratory will include the scientific name for each taxon in the macrofaunal abundance data submitted to the Project Manager.

Macrofaunal data will be analyzed for the following community parameters: abundance, Shannon-Wiener diversity index (H'), Pielou's evenness (J'), Margalef’s diversity index (DMg), Simpson, and/or total taxonomic distinctness.

Shannon-Wiener diversity index characterizes the species diversity in a community and is calculated following

H’ =

where *p*i is the proportion of individuals belonging to the *i*th species in the sample.

Pielou's evenness is calculated by

J’ = H’/H’max

Where H’ is derived from the Shannon-Weiner diversity index and H’max is the maximum possible value of H’ (if every species were equally likely), calculated by H’ = ln *S*. *S* is species richness, the total number of species in the sample. J’ ranges between 0 and 1, with lower numbers indicating less evenness between the species.

Margalef’s diversity index (DMg) is calculated by

DMg = (*S* - 1)/ln N

where *S* is species richness and N is the total number of individuals in the sample.

Total taxonomic distinctness can be calculated[[22]](#footnote-23) which can be used to document change in taxonomic distinctness with increased stress.[[23]](#footnote-24) Distinctness is different from species diversity in that it describes the phylogenetic distance between observed species, as well as the diversity of function. (See Warwick and Clarke 1999: <http://www.int-res.com/articles/meps/184/m184p021.pdf>).

The results of all statistical analyses will be combined and tabulated into an Excel spreadsheet for delivery to the Project Manager.

Insert Sections B10.2-4 from page 84

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === false +++

## B2 Benthic Sample Collection, Processing, and Storage Overview

Processing and storage requirements for sediment samples are described in the following sections.

### B2.1 Soft-Bottom Grab Sample Collection

A 0.04 m2 Ted Young-modified Van Veen grab sampler will be used to collect bottom sediment samples. At each station, one grab sample will be collected for total organic carbon and grain size analysis.

Supply list:

* Young-modified Van Veen grab with grab stand or frame if needed
* Weights and pads for grab
* Nitrile gloves
* Plastic tub or bucket
* Electrical tape
* Ruler (cm)
* Squirt bottle (ambient water)
* Stainless steel mixing pot or bowl with lid
* Stainless steel or Teflon spoons (15”), scoops, or spatula
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (500 mL) with screw-cap lids
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (125 mL) with screw-cap lids
* Scrub brush
* Cooler with wet ice (for grain size samples)
* Cooler with dry ice (for total organic carbon samples)

The following items will also be needed for recording measurements:

* Sampling log form
* Sample collection form
* Pencils
* Waterproof paper for internal sample jar labels
* Fine-tipped indelible markers (for labels)
* Write-on colored tape or pre-printed write-on labels
* Clear tape strips

Prior to sample collection (before the sample jar gets wet), an external label (including station location, replicate number, and date) will be taped to the outside of jars. The external label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Labels made of waterproof paper will be filled in onsite with a pencil.

Once the sampler is at the sampling site, and coordinates have been verified, the sediment grab will be deployed.

1. Attach the sampler to the end of the winch cable with a shackle and tighten the pin (or secure the pin with a cable tie).
2. Set the grab according to the manufacturer’s instructions and disengage any safety device designed to lock the sampler open.
3. Lower the grab sampler through the water column no faster than about 1 m/second. This minimizes the effects of wave disturbance on surficial sediments.
4. Allow a moment for the sampler to settle into the substrate and then allow slack on the cable (letting the cable go slack serves to release the jaws of the sampler so they will close as the sampler is retrieved).
5. Retrieve the sampler and lower it into its cradle or a plastic tub on the dock or pier. Open the top and determine whether the sampling is successful or not (see Figure A7.1).
6. A successful grab is one having relatively level, intact sediment over the entire area of the grab, and a sediment depth at the center of at least 7 cm for the benthic macroinvertebrate grab.
7. Grabs containing no sediment, partially filled grabs, or grabs with shelly/rocky substrates or grossly slumped surfaces are unacceptable.
8. Grabs completely filled to the top, where the sediment is in direct contact with the hinged top, are also unacceptable.
9. It may take several attempts using different amounts of weight to obtain the first acceptable sample. More weight will result in a deeper bite of the grab. In very soft mud, pads may be needed to prevent the sampler from sinking into the mud. If pads are used, the rate of descent near the bottom should be slowed even further to reduce the wave.
10. If, after several attempts, only grabs less than 7 cm deep can be obtained, use the next successful grab regardless of the depth of sediment at the center of the grab.
11. Use the comments section on the sample collectionform to describe sampling efforts and accurately record the depth of the sediment captured by the grab.
12. Carefully drain overlying water from the grab.
13. Enter notes on the condition of the sample (smell, substrate, presence of organisms on the surface, etc.) on the sample collectionform.
14. Process the grab sample for sediment testing as described below.

Precautions will be taken during the deployment and retrieval of the grab sampler to prevent contamination of samples between stations. Sampling for grain size and total organic carbon determinations requires that the grab and associated sampling equipment be washed and rinsed with screened ambient seawater until free of all sediment.

The penetration depth, sediment volume, and sediment texture and character will be visually estimated for acceptable samples. These data will be recorded in the field log.

#### Penetration Depth

The penetration depth of the sample will then be measured using a plastic ruler (marked in mm) pushed into the sediment. Any sediment adhering to the surface of the ruler will be rinsed back into the grab for processing with the remainder of the sample.

#### Sediment Volume

The volume of the grab will be estimated by comparing the measured penetration depth according to Table B2.1.

Table B2.1. Values Used to Convert Grab Penetration Depth to Sediment Volume

| Grab Penetration Depth (cm)1 | Sediment Volume (L) 0.04-m2 Grab |
| --- | --- |
| 4.1-5.0 | 1.4 |
| 5.1-6.0 | 1.8 |
| 6.1-7.0 | 2.1 |
| 7.1-8.0 | 2.4 |
| 8.1-9.0 | 2.7 |
| 9.1-10.0 | 3.0 |
| 10.1-11.0 |  |
| 11.1-12.0 |  |
| 12.1-13.0 |  |
| 13.1-14.0 |  |
| 14.1-15.0 |  |

1Over penetration is > 9.5 cm for 0.04-m2 grab.

#### Sediment Texture and Character

The sediment should be characterized as being coarse sand, fine sand, silt, clay, gravel, mud, or of a mixed type. The presence of shell hash should also be recorded. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

### B2.2 Sediment sample processing

#### Grain Size

If the grab sample to be used for sediment analysis meets the acceptability criteria, the water overlying the sample will be siphoned from the grab and the surface sediment (top 0 to 2 cm) will be collected with a scoop and transferred to a clean (rinsed with clean seawater) glass bowl. The sediment will be thoroughly homogenized before being transferred to 500 mL (16 oz) wide-mouth sample jars to hold approximately 500 mL subsamples for grain size analysis. These samples will be labeled and kept on ice at 1 to 4C for delivery to the laboratory within 24 hours of survey completion.

#### Total Organic Carbon

If the grab sample to be used for sediment analysis meets the acceptability criteria, the water overlying the sample will be siphoned from the grab and the surface sediment (top 0 to 2 cm) will be collected with a scoop and transferred to a clean (rinsed with clean seawater) glass bowl. The sediment will be thoroughly homogenized before being transferred to 125 mL (4 oz) wide-mouth sample jars to hold approximately 50 mL subsamples for total organic carbon analysis. These samples will be labeled and set on dry ice (frozen) for delivery to the contracted laboratory within 24 hours of survey completion.

### B2.3 Soft-Bottom Grab Sample Storage

The maximum holding time in the laboratory for grain size samples will be 28 days with refrigeration, and samples for total organic carbon analysis will be 28 days frozen. These time frames are consistent with a number of standard EPA Methods and ensure that samples are analyzed in a timely manner to prevent or minimize analyte degradation and interferences.

### B3 Sample Handling and Custody

### B3.1 Sample Handling

Handling of samples while in the field, including storage requirements, is described in Section B2 above.

Sediment chemistry samples must be kept cold (sediment grain size samples) or frozen (total organic carbon samples). After the survey is completed, a survey crew member will deliver the sediment chemistry samples to the contracted laboratory according to a pre-arranged schedule for sample dropoff. If circumstances dictate that the samples must be shipped to the laboratory, they will be shipped by overnight express. In that case, the samples that were frozen after collection will be placed on dry ice with protective layers of foam or bubble wrap to ensure that they remain intact and frozen during shipment.

### B3.2 Sample Custody

#### Sample Tracking

Sample custody will be tracked through external and internal sample labels, sample collection forms, and chain of custody forms.

Sediment samples collected under this QAPP will be processed by a contracted laboratory. The contracted laboratory will provide the sample containers and sample labels. Sample labels will contain or have spaces for following information: station location, survey type, analysis, preservative, date/time collected, and collector’s name. The Chief Scientist is responsible for verifying that information on the sample labels matches the information on the chain of custody forms before delivering the samples to the contracted laboratory.

The survey crew will fill out the sample log at each station. The log includes header fields for entering pertinent information about each station, such as arrival time, bottom depth, and weather observations. The log contains spaces for specific grab data, such as penetration depth and general descriptions. These records will remain in the survey logbook and will be maintained in the project files. During field collection, chain of custody forms also will be completed. The chain of custody forms will include the unique information from the corresponding label on the sample container, ensuring the tracking of sample location and status.

#### Sample Custody

Sediment samples will be in the custody of the survey Chief Scientist or a crew member from collection until they are transferred to the contracted laboratory. Transfer of sediment samples will be documented on the custody forms. All samples will be distributed to the appropriate laboratory personnel by hand or by a shipping service. The field sample custodian will retain a copy of the chain of custody form in the field log. The original will accompany the samples to the laboratory for subsequent sample transfer. When samples arrive at the laboratory, custody will be relinquished to the laboratory staff. The laboratory staff will verify that the custody seals on the cooler are intact. The laboratory staff will then examine the samples, verify that sample-specific information recorded on the chain of custody form is accurate and that the sample integrity is uncompromised, log the samples into their laboratory tracking system, and complete and sign the chain of custody form so that transfer of custody of the samples is complete. Any discrepancies between sample labels and transmittal forms, the condition of the samples upon receipt, and any unusual events or deviations from the QAPP will be documented in detail on the chain of custody form, and the Project Manager notified. Copies of completed custody forms will be delivered (scanned and emailed or faxed) to the Project Manager within 24 hours of receipt.

## B4 Analytical Methods

Sediment geophysical properties, including sediment grain size and total organic carbon, are based on laboratory assessment of soft-bottom grab samples.

Table B4.1 Marine Benthic Survey Sample Analyses, Sediment

| **Parameter** | **Unit of Measurement** | **Method** | **Reference** |
| --- | --- | --- | --- |
| TOC | %C by dry weight | Lloyd Kahn | Kahn, 1988[[24]](#footnote-25) |
| Sediment Grain Size | % dry weight | Folk, 1974[[25]](#footnote-26)  FGDC, 2012[[26]](#footnote-27) | Sweeny and Rutecki, 2019[[27]](#footnote-28) |

## B.5 Quality Control

### B5.1 Sediment Samples Quality Control

#### Accuracy, Precision, and Representativeness

These qualities will be assured by compliance with the sampling plan and by ensuring that samples are well homogenized and subsampled and preserved following methods detailed in Section B2.

#### Comparability

Procedures for sampling and subsampling are comparable to those used in other investigations in Massachusetts coastal waters.

#### Completeness

All required samples will be collected at all of the stations specified in the program sampling plan.

### B5.2 Soft-Bottom Grab Sampling Quality Control

All sediment samples to be used for grain size and total organic carbon analysis will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment
* Control of penetration by adding or removing weights to the frame and adjusting descent rate
* Slow recovery until the grab is free of the bottom
* Inspection for signs of leakage
* Securing the grab on deck

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* The sampler is not overfilled with sediment; the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors.
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable if:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside that grid
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

No analytical laboratory instruments are covered by this QAPP.

## B7 Instruments

No analytical laboratory instruments are covered by this QAPP.

Insert Section B8 from page 82

Insert Section B9 from page 83

## B10 Data Management—Sediment Analysis

### B10.1 Sediment Analysis

The contracted laboratory will include sediment grain size and percentage of total organic carbon in the sediment data submitted to the Project Manager. After data verification, sediment samples may be disposed of following internal laboratory protocols. Sediment samples with known toxins (e.g., PCBs, dioxin, and PAHs) will be disposed of properly following local, state, and federal laws.

Insert Sections B10.2-4 from page 84

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === false +++

## B2 Benthic Sample Processing and Storage Overview

Processing and storage requirements for sediment samples are described in the following sections.

### B2.1 Soft-Bottom Grab Sample Collection

A 0.04 m2 Ted Young-modified Van Veen grab sampler will be used to collect bottom sediment samples. At each station, one grab sample will be collected for grain size analysis.

Supply list:

* Young-modified Van Veen grab with grab stand or frame if needed
* Weights and pads for grab
* Nitrile gloves
* Plastic tub or bucket
* Electrical tape
* Ruler (cm)
* Squirt bottle (ambient water)
* Stainless steel mixing pot or bowl with lid
* Stainless steel or Teflon spoons (15”), scoops, or spatula
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (500 mL) with screw-cap lids
* Plastic bags (e.g., Whirl Pak, for grain size samples)
* Scrub brush
* Cooler with wet ice

The following items will also be needed for recording measurements:

* Sampling log form
* Sample collection form
* Pencils
* Waterproof paper for internal sample jar labels
* Fine-tipped indelible markers (for labels)
* Write-on colored tape or pre-printed write-on labels
* Clear tape strips

Prior to sample collection (before the sample jar gets wet), an external label (including station location, replicate number, and date) will be taped to the outside of jars. The external label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Labels made of waterproof paper will be filled in onsite with a pencil.

Once the sampler is at the sampling site, and coordinates have been verified, the sediment grab will be deployed.

1. Attach the sampler to the end of the winch cable with a shackle and tighten the pin (or secure the pin with a cable tie).
2. Set the grab according to the manufacturer’s instructions and disengage any safety device designed to lock the sampler open.
3. Lower the grab sampler through the water column no faster than about 1 m/second. This minimizes the effects of bow wave disturbance on surficial sediments.
4. Allow a moment for the sampler to settle into the substrate and then allow slack on the cable (letting the cable go slack serves to release the jaws of the sampler so they will close as the sampler is retrieved).
5. Retrieve the sampler and lower it into its cradle or a plastic tub on the dock or pier. Open the top and determine whether the sampling is successful or not (see Figure 1).
6. A successful grab is one having relatively level, intact sediment over the entire area of the grab, and a sediment depth at the center of at least 7 cm for the benthic macroinvertebrate grab.
7. Grabs containing no sediment, partially filled grabs, or grabs with shelly/rocky substrates or grossly slumped surfaces are unacceptable.
8. Grabs completely filled to the top, where the sediment is in direct contact with the hinged top, are also unacceptable.
9. It may take several attempts using different amounts of weight to obtain the first acceptable sample. More weight will result in a deeper bite of the grab. In very soft mud, pads may be needed to prevent the sampler from sinking into the mud. If pads are used, the rate of descent near the bottom should be slowed even further to reduce the bow wave.
10. If, after several attempts, only grabs less than 7 cm deep can be obtained, use the next successful grab regardless of the depth of sediment at the center of the grab.
11. Use the comments section on the sample collectionform to describe sampling efforts and accurately record the depth of the sediment captured by the grab.
12. Carefully drain overlying water from the grab.
13. Enter notes on the condition of the sample (smell, substrate, presence of organisms on the surface, etc.) on the sample collectionform.
14. Process the grab sample for sediment testing as described below.

Take precautions during the deployment and retrieval of the grab sampler to prevent contamination of samples between stations. Sampling for grain size determinations requires that the grab and associated sampling equipment be washed and rinsed with screened ambient seawater until free of all sediment.

The penetration depth, sediment volume, and sediment texture will be visually estimated for acceptable samples. These data will be recorded in the field log. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

#### Penetration Depth

The penetration depth of the sample will then be measured using a plastic ruler (marked in mm) pushed into the sediment. Any sediment adhering to the surface of the ruler will be rinsed back into the grab for processing with the remainder of the sample.

#### Sediment Volume

The volume of the grab will be estimated by comparing the measured penetration depth according to Table B2.1.

Table B2.1. Values Used to Convert Grab Penetration Depth to Sediment Volume

| Grab Penetration Depth (cm)1 | Sediment Volume (L) 0.04-m2 Grab |
| --- | --- |
| 4.1-5.0 | 1.4 |
| 5.1-6.0 | 1.8 |
| 6.1-7.0 | 2.1 |
| 7.1-8.0 | 2.4 |
| 8.1-9.0 | 2.7 |
| 9.1-10.0 | 3.0 |
| 10.1-11.0 |  |
| 11.1-12.0 |  |
| 12.1-13.0 |  |
| 13.1-14.0 |  |
| 14.1-15.0 |  |

1Over penetration is > 9.5 cm for 0.04-m2 grab.

#### Sediment Texture and Character

The sediment should be characterized as being coarse sand, fine sand, silt, clay, gravel, mud, or of a mixed type. The presence of shell hash should also be recorded. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

#### Sediment Analysis Sample Processing for Grain Size

If the grab sample to be used for sediment analysis meets the acceptability criteria, the water overlying the sample will be siphoned from the grab and the surface sediment (top 0 to 2 cm) will be collected with a scoop and transferred to a clean (rinsed with clean seawater) glass bowl. The sediment will be thoroughly homogenized before being transferred to 500 mL (16 oz) wide-mouth sample jars to hold approximately 500 mL subsamples for grain size analysis. These samples will be labeled and kept on ice at 1 to 4C for delivery to the laboratory within 48 hours of survey completion.

The maximum holding time for sediment samples in the laboratory will be 28 days with refrigeration. This time frame is consistent with a number of standard EPA Methods and ensures that samples are analyzed in a timely manner to prevent or minimize analyte degradation and interferences.

## B3 Sample Handling and Custody

Section B2 describes handling of samples while in the field, including storage requirements.

The sediment chemistry samples collected during the benthic survey must be kept cold as described in Table 8. After the survey is completed, a survey crew member will deliver the sediment chemistry samples to the contracted laboratory according to a pre-arranged schedule for sample dropoff. If circumstances dictate that the samples must be shipped to the laboratory, they will be shipped by overnight express.

### B3.2 Sample Custody

#### Sample Tracking

Sample custody will be tracked through external and internal sample labels, sample collection forms, and chain of custody forms.

Sediment samples collected under this QAPP will be processed by a contracted laboratory. The contracted laboratory will provide the sample containers and sample labels. Sample labels will contain or have spaces for following information: station location, survey type, analysis, preservative, date/time collected, and collector’s name. The Chief Scientist is responsible for verifying that sample information on the sample labels matches the information on the chain of custody forms before delivering the samples to the contracted laboratory.

The survey crew will fill out the sample log at each station. The log includes headers for entering pertinent information about each station, such as arrival time, bottom depth, and weather observations. In addition, the log contains spaces for specific grab data, such as penetration depth and general descriptions. These records will remain in the survey logbook and will be maintained in the project files. During field collection, chain of custody forms also will be completed. The chain of custody forms will include the unique information from the corresponding label on the sample container, ensuring the tracking of sample location and status.

#### Sample Custody

Sediment samples will be in the custody of the survey Chief Scientist or a crew member from collection until they are transferred to the contracted laboratory. Transfer of sediment samples will be documented on the custody forms. All samples will be distributed to the appropriate laboratory personnel by hand or by a shipping service. The field sample custodian will retain a copy of the chain of custody form in the field log. The original will accompany the samples to the laboratory for subsequent sample transfer. When samples arrive at the laboratory, custody will be relinquished to the laboratory staff. The laboratory staff will verify that the custody seals on the cooler are intact. The laboratory staff will then examine the samples, verify that sample-specific information recorded on the chain of custody form is accurate and that the sample integrity is uncompromised, log the samples into their laboratory tracking system, and complete and sign the chain of custody form so that transfer of custody of the samples is complete. Any discrepancies between sample labels and transmittal forms, the condition of the samples upon receipt, and any unusual events or deviations from the QAPP will be documented in detail on the chain of custody form, and the Project Manager notified. Copies of completed custody forms will be delivered (scanned and emailed or faxed) to the Project Manager within 24 hours of receipt.

## B5 Sample Quality Control

### B5.1 Sediment Sample Quality Control

#### Accuracy, Precision, and Representativeness

These qualities will be assured by the sampling plan and by ensuring that samples are well homogenized and subsampled and preserved following methods detailed in Section B2.

#### Comparability

Procedures for sampling and subsampling are comparable to those used in other investigations in Massachusetts coastal waters.

#### Completeness

All required samples will be collected at all of the stations specified in the embayment-specific study plan.

### B5.2 Soft-Bottom Grab Sampling Quality Control

All sediment samples to be used for grain size analysis will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment
* Control of penetration by adding or removing weights to the frame and adjusting descent rate
* Slow recovery until the grab is free of the bottom
* Inspection for signs of leakage
* Securing the grab on deck

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* The sampler is not overfilled with sediment; the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors.
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable if:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside that grid.
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

No analytical laboratory instruments are covered by this QAPP.

## B7 Instruments

No analytical laboratory instruments are covered by this QAPP.

Insert Section B8 from page 82 above

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other studies water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records.
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments).

Secondary data used will be documented in the Secondary Data Form, attached, according to Sections A9 and C2.

Insert Section B9 from page 83 above

## B10 Data Management

### B10.1 Sediment Analysis

The contracted laboratory will include sediment grain size in the sediment data submitted to the Project Manager. After data verification, sediment samples can be disposed of following internal laboratory protocols. Sediment samples with known toxins (e.g., PCBs, dioxin, and PAHs) will be disposed of properly following local, state, and federal laws.

Insert Sections B10.2-4 from page 84

+++END-IF+++

+++IF determine('Saltwater Benthic', 'Saltwater' ,' Sample penetration depth', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grab sample volume', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Sediment texture', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,' Grain size', 'Sediment grab samples') === false && determine('Saltwater Benthic', 'Saltwater' ,' Total organic carbon (TOC)', 'Sediment grab samples') === true && determine('Saltwater Benthic', 'Saltwater' ,'Infauna', 'Sediment grab samples') === false +++

## B2 Benthic Sample Processing and Storage

Processing and storage requirements for sediment samples are described in the following sections.

### B2.1 Soft-Bottom Grab Sample Collection

A 0.04 m2 Ted Young-modified Van Veen grab sampler will be used to collect bottom sediment samples. At each station, one grab sample will be collected for total organic carbon analysis.

Supply list:

* Young-modified Van Veen grab with grab stand or frame if needed
* Weights and pads for grab
* Nitrile gloves
* Plastic tub or bucket
* Electrical tape
* Ruler (cm)
* Squirt bottle (ambient water)
* Stainless steel mixing pot or bowl with lid
* Stainless steel or Teflon spoons (15”), scoops, or spatula
* Glass or Nalgene (or other sturdy plastic) wide-mouth sample jars (125 mL) with screw-cap lids
* Scrub brush
* Cooler with dry ice

The following items will also be needed for recording measurements:

* Sampling log form
* Sample collection form
* Pencils
* Waterproof paper for internal sample jar labels
* Fine-tipped indelible markers (for labels)
* Write-on colored tape or pre-printed write-on labels
* Clear tape strips

Prior to sample collection (before the sample jar gets wet), an external label (including station location, replicate number, and date) will be taped to the outside of jars. The external label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Labels made of waterproof paper will be filled in onsite with a pencil.

Once the sampler is at the sampling site, and coordinates have been verified, the sediment grab will be deployed.

1. Attach the sampler to the end of the winch cable with a shackle and tighten the pin (or secure the pin with a cable tie).
2. Set the grab according to the manufacturer’s instructions and disengage any safety device designed to lock the sampler open.
3. Lower the grab sampler through the water column no faster than about 1 m/second. This minimizes the effects of wave disturbance on surficial sediments.
4. Allow a moment for the sampler to settle into the substrate and then allow slack on the cable (letting the cable go slack serves to release the jaws of the sampler so they will close as the sampler is retrieved).
5. Retrieve the sampler and lower it into its cradle or a plastic tub on the dock or pier. Open the top and determine whether the sampling is successful or not (see Figure A7.1).
6. A successful grab is one having relatively level, intact sediment over the entire area of the grab, and a sediment depth at the center of at least 7 cm for the benthic macroinvertebrate grab.
7. Grabs containing no sediment, partially filled grabs, or grabs with shelly/rocky substrates or grossly slumped surfaces are unacceptable.
8. Grabs completely filled to the top, where the sediment is in direct contact with the hinged top, are also unacceptable.
9. It may take several attempts using different amounts of weight to obtain the first acceptable sample. More weight will result in a deeper bite of the grab. In very soft mud, pads may be needed to prevent the sampler from sinking into the mud. If pads are used, the rate of descent near the bottom should be slowed even further to reduce the bow wave.
10. If, after several attempts, only grabs less than 7 cm deep can be obtained, use the next successful grab regardless of the depth of sediment at the center of the grab.
11. Use the comments section on the sample collectionform to describe sampling efforts and accurately record the depth of the sediment captured by the grab.
12. Carefully drain overlying water from the grab.
13. Enter notes on the condition of the sample (smell, substrate, presence of organisms on the surface, etc.) on the sample collectionform.
14. Process the grab sample for sediment testing as described below.

Take precautions during the deployment and retrieval of the grab sampler to prevent contamination of samples between stations. Sampling for total organic carbon determination requires that the grab and associated sampling equipment be washed and rinsed with screened ambient seawater until free of all sediment.

The penetration depth, sediment volume, and sediment texture and character will be visually estimated for acceptable samples. These data will be recorded in the field log.

#### Penetration Depth

The penetration depth of the sample will then be measured using a plastic ruler (marked in mm) pushed into the sediment. Any sediment adhering to the surface of the ruler will be rinsed back into the grab for processing with the remainder of the sample.

#### Sediment Volume

The volume of the grab will be estimated by comparing the measured penetration depth according to Table B2.1.

Table B2.1. Values Used to Convert Grab Penetration Depth to Sediment Volume

| Grab Penetration Depth (cm)1 | Sediment Volume (L) 0.04-m2 Grab |
| --- | --- |
| 4.1-5.0 | 1.4 |
| 5.1-6.0 | 1.8 |
| 6.1-7.0 | 2.1 |
| 7.1-8.0 | 2.4 |
| 8.1-9.0 | 2.7 |
| 9.1-10.0 | 3.0 |
| 10.1-11.0 |  |
| 11.1-12.0 |  |
| 12.1-13.0 |  |
| 13.1-14.0 |  |
| 14.1-15.0 |  |

1Over penetration is > 9.5 cm for 0.04-m2 grab.

#### Sediment Texture and Character

The sediment should be characterized as being coarse sand, fine sand, silt, clay, gravel, mud, or of a mixed type. The presence of shell hash should also be recorded. Obvious odors such as hydrogen sulfide (the odor of rotten eggs) or petroleum, or a lack of noticeable odors, should be recorded. General sediment colors (i.e., black, green, brown, red, or gray) should also be recorded.

#### Sediment Analysis Sample Processing for Total Organic Carbon

If the grab sample to be used for sediment analysis meets the acceptability criteria, the water overlying the sample will be siphoned from the grab and the surface sediment (top 0 to 2 cm) will be collected with a scoop and transferred to a clean (rinsed with clean seawater) glass bowl. The sediment will be thoroughly homogenized before being transferred to 125 mL (4 oz) wide-mouth sample jars to hold approximately 50 mL subsamples for total organic carbon analysis. These samples will be labeled and set on dry ice (frozen). These samples will be delivered to the contracted laboratory for analysis using the Lloyd Khan method within 24 hours of survey completion.

The maximum holding time for sediment samples in the laboratory will be 28 days frozen. This time frame is consistent with a number of standard EPA Methods and ensures that samples are analyzed in a timely manner to prevent or minimize analyte degradation and interferences.

## B3 Sample Handling and Custody

### B3.1 Sample Handling

Handling of samples while in the field, including storage requirements, is described in Section B2.

The sediment chemistry samples collected during the benthic survey must be kept frozen. After the survey is completed, a survey crew member will deliver the sediment chemistry samples to the contracted laboratory according to a pre-arranged schedule for sample dropoff. If circumstances dictate that the samples must be shipped to the laboratory, they will be shipped by overnight express. In that case, the samples that were frozen after collection will be placed on dry ice with protective layers of foam or bubble wrap to ensure that they remain intact and frozen during shipment.

### B3.2 Sample Custody

#### Sample Tracking

Sample custody will be tracked through external and internal sample labels, sample collection forms, and chain of custody forms.

Sediment samples collected under this QAPP will be processed by a contracted laboratory. The contracted laboratory will provide the sample containers and sample labels. Sample labels will contain or have spaces for following information: station location, survey type, analysis, preservative, date/time collected, and collected by. The Chief Scientist is responsible for verifying sample information on the sample labels matches the information on the chain of custody forms prior to delivering the samples to the contracted laboratory.

The survey crew will fill out the sample log at each station. The log includes header fields for entering pertinent information about each station, such as arrival time, bottom depth, and weather observations. In addition, the log contains spaces for specific grab data, such as penetration depth and general descriptions. These records will remain in the survey logbook and will be maintained in the project files. During field collection, chain of custody forms also will be completed. The chain of custody forms will include the unique information from the corresponding label on the sample container, ensuring the tracking of sample location and status.

#### Sample Custody

Sediment samples will be in the custody of the survey Chief Scientist or a crew member from collection until they are transferred to the contracted laboratory. Transfer of sediment samples will be documented on the custody forms. All samples will be distributed to the appropriate laboratory personnel by hand or by a shipping service. The field sample custodian will retain a copy of the chain of custody form in the field log. The original will accompany the samples to the laboratory for subsequent sample transfer. When samples arrive at the laboratory, custody will be relinquished to the laboratory staff. The laboratory staff will verify that the custody seals on the cooler are intact. The laboratory staff will then examine the samples, verify that sample-specific information recorded on the chain of custody form is accurate and that the sample integrity is uncompromised, log the samples into their laboratory tracking system, and complete and sign the chain of custody form so that transfer of custody of the samples is complete. Any discrepancies between sample labels and transmittal forms, the condition of the samples upon receipt, and any unusual events or deviations from the QAPP will be documented in detail on the chain of custody form, and the Project Manager notified. Copies of completed custody forms will be delivered (scanned and emailed or faxed) to the Project Manager within 24 hours of receipt.

## B5 Sample Quality Control

### B5.1 Sediment Samples Quality Control

#### Accuracy, Precision, and Representativeness

These qualities will be assured by compliance with the sampling plan and ensuring that samples are well homogenized and subsampled and preserved following methods detailed in Section B2.

#### Comparability

Procedures for sampling and subsampling are comparable to those used in other investigations in Massachusetts coastal waters.

#### Completeness

All required samples will be collected at all of the stations specified in the program sampling plan.

### B5.2 Soft-Bottom Grab Sampling Quality Control

All sediment samples to be used for total organic carbon analysis will be collected with a 0.04 m2 Young-modified Van Veen grab sampler. Samples will be kept undisturbed through careful attention to established deployment and recovery procedures. Procedures used by survey crews will cover the following aspects of deployment and recovery:

* Thorough wash-down of the grab before each deployment
* Control of penetration by adding or removing weights to the frame and adjusting descent rate
* Slow recovery until the grab is free of the bottom
* Inspection for signs of leakage
* Securing the grab on deck

Each grab sample will be inspected for signs of disturbance. The following criteria identify ideal characteristics for an acceptable grab sample:

* The sampler is not overfilled with sediment; the sediment surface is intact and relatively level over the entire area of the grab. The jaws must be fully closed and the top of the sediment must be below the level of the opening doors.
* Overlying water is present and not excessively turbid.
* Sediment depth at the center of the sampler is at least 7 cm, indicating that the desired penetration was achieved.

Mild overfill may be acceptable if:

* The sediment surface is intact.
* There is no evidence that the surface sediment has pushed through the grid surface of the grab—i.e., no visible imprint from the screening outside that grid.
* There is no evidence that sediment has pushed out through the hinge or the edges of the grab.

The overall condition of the grab will be documented on the station log.

## B6 Instrument/Equipment Testing, Inspection, and Maintenance Requirements

No analytical laboratory instruments are covered by this QAPP.

## B7 Instruments

No analytical laboratory instruments are covered by this QAPP.

Insert Section B8 from page 82 above

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other studies water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records.
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments).

Secondary data used will be documented in the Secondary Data Form, attached, according to Sections A9 and C2.

Insert Section B9 from page 83 above

## B10 Data Management

### B10.1 Sediment Analysis

The contracted laboratory will include sediment grain size in the sediment data submitted to the Project Manager. After data verification, sediment samples can be disposed of following internal laboratory protocols. Sediment samples with known toxins (e.g., PCBs, dioxin, and PAHs) will be disposed of properly following local, state, and federal laws.

Insert Sections B10.2-4 from page 84

+++END-IF+++

+++END-IF+++

+++IF determine('Saltwater Water Quality','Saltwater','','') === true+++

# Section B. Marine/Water Quality Data Generation And Acquisition

## B1 Sampling Design

### B1.1 Sampling Site Selection – AquaQAPP concern = source impact

For point source assessments, the sampling site selection will include stations upstream and downstream of the source, as well as reference stations. The site selected will be in an area where the effluent has had a reasonable opportunity to mix. In cases where a mixing zone has been defined in a license, sampling will be done immediately downstream of it. Where the effluent plume channels down one bank for great distances (over 1 km), or where localized effluent impact is expected to be severe for a distance beyond the zone of initial dilution, sampling at a site upstream of the source, one or more in the plume, and at least two farther downstream will take place. One downstream site will be located at the point of presumed bank-to-bank mixing; subsequent sites will be chosen to assess the extent of impact downstream. The area of initial dilution of an effluent will be determined by visual observation of the plume pattern; by observations of biotic effects attributable to the plume, if evident (benthic algal biomass growth, die-off patterns); and by transects of conductivity (specific conductance) measurements from the outfall, in a downstream direction.

Sampling sites will be selected to ensure that the physical characteristics among sampling sites are similar. For surveys conducted to determine use designations, sampling locations will be representative of the stream reach. Reference conditions will include minimally impaired sites in the same ecoregion, size class, and stream type (width, depth, gradient).

### B1.1 Sampling Site Selection – AquaQAPP concern = general coastal water health

For water quality condition assessment, routine sampling activities will consist of collecting instream samples. Routine sampling will be representative of overall water quality and are relatively unchanging over time to allow comparison to past and future investigations. Sites will be generally selected at the downstream ends and/or key segmentation points of major tributaries and at or near locations where there is a longstanding data record.

To meaningfully evaluate water quality condition, sampling locations will be selected to ensure generally comparable physical habitat. Sample locations will be scouted out ahead of time to identify appropriate reaches and accessibility for sampling. Station siting of both study sites and reference sites will take place during June. Reconnaissance activities prior to June are not desirable, as instream conditions—most notably flow regimes—during this time are often dramatically different than during the sampling index period. To lay out the sampling reach and sites, the route to the site will be explored to ensure it is free of obstacles that would prohibit sampling and data collection activities, then the sample reach characteristics will be assessed. Field conditions (e.g., instream and riparian habitat characteristics, surrounding land use, observations of nonpoint source pollution or other pertinent information) during the time of reconnaissance will be noted and recorded in a field notebook.

## B2 Sampling Methods: Sample Collection, Processing, and Storage

### B1.3 Sample Collection Methods

Sample types include grab samples and direct measurements using electronic instruments in the field. Water quality parameters that are measured/observed in situ as well as indicators to be analyzed in the laboratory are listed in Table 1.2.

Table B1.1. Marine Field Sampling Summary

| Parameter - Method | Number of sampling locations | 1Rationale for number of samples | 2Site location rationale | Frequency | Number/type of QC samples including field duplicates (10%) and blanks (10%) |
| --- | --- | --- | --- | --- | --- |
| +++FOR parameter IN sampleDesign.filter((param) => param.monitoringCategory === 'Saltwater Water Quality') +++ |  |  |  |  |  |
| +++ **INS $**parameter.sampleParameter+++ | +++ **INS $**parameter.numSampleLocations+++ | +++ **INS $**parameter.sampleNumRationale+++ | +++ **INS $**parameter.locationRationale+++ | +++ **INS $**parameter.frequency+++ | +++ **INS $**parameter.numQcSamples+++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

**1Dropdown:**

* Random or probabilistic
* Accessibility considerations
* Proximity to potential pollutant source
* Replication of previous sampling efforts (*e.g.*, by DEP or EPA)
* Other (please specify): \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

**2Dropdown:**

* Spatial coverage of waterbody
* Feature of interest
* Regulatory requirement
* Proximity to impact or suspected pollution source
* Capacity (funding or staffing) Replication of previous sampling efforts (*e.g.*, by DEP or EPA)
* Other (please specify): \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

Table B2.1. Equipment Preparation, Sample Processing, and Storage Requirements

| Parameter | Sample collection method | Container Type and Preparation | Minimum Sample Quantity | Sample Preservation | Maximum Holding Time |
| --- | --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality')+++ |  |  |  |  |  |
| +++ **INS $**parameter.label+++ | +++ **INS $**parameter.sampleCollectionMethod+++ | +++ **INS $**parameter.sampleContainer+++ | +++ **INS $**parameter.sampleVolume+++ | +++ **INS $**parameter.samplePreservation+++ | +++ **INS $**parameter.maxHoldingTime +++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

Prior to sample collection, sample bottle labels (including station location, replicate number, and date) will be taped to the outside of sample containers. The labels may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling measuring or collection device will be deployed.

### B2.1 In Situ Water Quality Monitoring

#### Equipment/Instrument Calibration

Prior to field use, the multi-parameter or individual units will be calibrated in accordance with the manufacturer’s instruction manual. If no instructions specific to an instrument are available, the following general calibration methods will be followed.

##### General Calibration Methods: For Multi-Parameter Unit or Individual Units

Supply list for taking measurements and calibrating the water quality sensor:

* Multi-parameter water quality sensor (with cable and handheld data logger) with pH, dissolved oxygen, temperature, and conductivity probes
* Extra batteries
* De-ionized water (lab certified preferred, but not required)
* Calibration cups and standards
* Barometer to use for calibration
* Secchi disk (20 cm diameter, weighted) and 100 ft line with clip (marked in 0.5 m intervals)
* Calibration records form

Calibration standards:

* pH 7.00 standard buffer solution
* pH 4.00 standard buffer solution
* pH 10.00 standard buffer solution
* 1 mS/cm (1,000 μS/cm) conductivity standard
* Sodium sulfite solution (0% dissolved oxygen)

The following items will also be needed for recording measurements:

* Field Data Fpr,
* Pencils (for data forms)

##### Equipment Calibration Method: Multi-Parameter Sonde or Individual sensors

###### Temperature sensor

Check the accuracy of the sensor against a thermometer that is traceable to the National Institute of Standards and Technology at least once per sampling season.

###### pH sensor

*Calibration standards required:* pH 4.00, 7.00 and 10.00 standard buffer solutions.

Calibrate the pH sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions and existing standard operating procedures. Ideally, use a QC solution that is similar in ionic strength to the water samples you will be measuring.

*Calibration method:*

1. Use the small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with Kimwipes or paper towel.
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration on the instrument calibration form.

Record the calibration solution lot number and expiration date on the instrument calibration form.

###### Dissolved Oxygen sensor

*Calibration standard required:* Sodium sulfite solution (0% dissolved oxygen).

Calibrate the dissolved oxygen unit before each sampling event. It is recommended that the sensor probe be calibrated in the field against an atmospheric standard (e.g., ambient air saturated with water). Follow your manufacturer’s guidelines for calibration of the dissolved oxygen probe.

*General calibration method:*

1. Use the small plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with Kimwipes or paper towel. (Never touch the membrane of the dissolved oxygen sensor.)
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration on the instrument calibration form.

###### Salinity sensor

Calibrate the salinity sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions. Ideally, a QC solution should be used that incorporates the entire conductivity range encountered in the NRSA and a record of test results kept on file.

###### Turbidity

*Calibration standard required:* TO BE PROVIDED

Calibrate the turbidity sensor prior to each sampling event. Calibrate the sensor in accordance with the manufacturer’s instructions.

*General Calibration method:*

1. Use plastic cups (washed and rinsed with distilled water) to hold calibration solutions during calibration.
2. Use calibration solutions at room temperature.
3. Ensure that the sensor being calibrated and the temperature probe are immersed in the calibrating solution during calibration.
4. Between calibration steps, rinse the sensors with ambient temperature distilled water, then gently blot with Kim-wipes or paper towel. (Never touch the membrane of the dissolved oxygen sensor.)
5. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration.
6. Record the calibration solution Lot Number and expiration date on the Instrument Calibration Log.

###### Multi-*Parameter Unit*

Calibrate the unit prior to each sampling event by following the user instruction manual and relevant standards applicable to the sensors/parameters on the unit. For each parameter, record the initial reading in the standard solution (before calibration) and the reading after calibration on the instrument calibration form. Record the calibration solution lot number and expiration date on the instrument calibration form.

#### Multi-Parameter Unit Deployment and Grab Sample Collection

Measurements of the parameters are generally taken at near-surface (0.1 m below the surface). If the monitoring program requires measurement of conditions in the water column, a hydrographic profile at each site will be obtained at water depth of grater than or equal to 2 meters. The parameters are measured to detect extremes in conditions that might indicate impairment and depth at location. In situ measurements will be made using a calibrated water quality multi‐parameter unit at each station. Measurements will be then collected as the sensor is lowered, at prescribed intervals (usually 0.5 m to 1.0 m depending on depth) down to 0.5 m from the bottom.

The total water depth at the sampling site is estimated by lowering the depth sounding line (marked in ft) to the bottom of the river and counting the number of taped 1 ft marks on the cable. If possible, sensor measurements will be collected during the downcast from near surface (approximately 0.5–1.5 m) to near bottom (about 0.5 m off the bottom) or along a hydrographic profile.

#### Method: In Situ Sampling Procedures Using a Calibrated Multi-Parameter Unit

If taking measurements offshore using a boat, locate the station using a GPS. When close to the station, position the vessel slightly up-current or upwind, with the side from which the unit is deployed facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the unit is deployed.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Multi-parameter unit
* Extra batteries

For recording measurements:

* Sample collection form
* Field Data Forms
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

1. Turn on the unit manually and lower over the side until the sensors are approximately 10 cm below the surface. Allow the sensors to equilibrate for at least 60 seconds.
2. Measure the total water depth at the station location to the nearest 0.1 m and record it on the Field Data Form. If the unit is attached to a data recorder, save hydrographic profile data as an electronic file.
3. Lower the unit into the water and record dissolved oxygen, pH, salinity/conductivity, and temperature measurements at the following depths: 0.1 m below the surface, 0.5 m below the surface, every 1 m from depths of 1.0 to 10.0 m, and (if the site is deeper than 10 m) every 5 m thereafter. Take the last set of measurements at 0.5 m from the bottom, making sure not to let the unit touch the bottom. Record these results in the downcast section of the Field Data Form.
4. Repeat the full sets of measurements at each of the same depth intervals as the probe is retrieved (upcast). Record all data on the Field Data Form.

After all in situmeasurements have been completed for the sampling day, perform a post-measurement calibration check of the pH and conductivity probes. Record these values on the instrument calibration form.

### B2.2 Alkalinity

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (With this setup, the boat’s expected drift will bring it to the station coordinates when the unit is deployed.) Deploy the anchor. Once the boat settles, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 1 L sample container
* Cooler with ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Field Data Forms
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

Method (using a sample bottle):

1. Attach a label to a 1 L HDPE bottle and cover it with clear plastic tape.
2. Put on nitrile gloves.
3. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap.
4. Rinse the bottle three times with water from one side of the boat.
5. Hold the bottle near its base and plunge it (opening downward) below the water surface on the opposite side of the boat. Collect a water sample 0.1 m beneath the surface.
6. Leave a 1 in. air space. Do not fill the bottle completely (so that the sample can be shaken just before analysis). Recap the bottle carefully, remembering not to touch the inside.
7. Place the bottle on ice in the cooler.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn bottle):

1. Attach a label to a 1 L HDPE bottle and cover it with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the HDPE bottle with sample water.
6. Replace the lid. Seal the lid sample bottle tightly with electrical tape.
7. Place the bottle in the cooler on ice.

Record the collection data on the sample collection form.

1. +++IF determine('Saltwater Water Quality','Saltwater','Turbidity','EPA 180.1') === true || determine('Saltwater Water Quality','Saltwater','Turbidity','SM 2130-B') === true || determine('Saltwater Water Quality','Saltwater','Total suspended solids','') === true || determine('Saltwater Water Quality','Saltwater','Water transparency (Secchi depth)','') === true +++

### B2.3 Turbidity

1. +++IF determine('Saltwater Water Quality','Saltwater','Turbidity','EPA 180.1') === true || determine('Saltwater Water Quality','Saltwater','Turbidity','SM 2130-B') === true +++

This section describes the procedures and methods for the field collection and preservation of the water samples to measure turbidity. Prior to sample collection, a label (including station location, replicate number, and date) will be taped to the outside of sample containers. The label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling collection device will be deployed.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 1 L sample container
* Cooler with ice
* De-ionized water

For recording measurements:

* Sample collection form
* Field Data Forms
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

Method (using a sample bottle):

1. Attach a label to 1 L HDPE bottle and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap.
4. Rinse the HDPE bottle three times with site water.
5. Hold the bottle near its base and plunge it (opening downward) below the water surface on the opposite side of the boat. Collect a water sample 0.1 m beneath the surface.
6. Recap the bottle carefully, remembering not to touch the inside.
7. Place the bottle in the cooler on ice.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn bottle):

1. Attach a label to a 1 L HDPE bottle and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the HDPE bottle with sample water.
6. Replace the cap and seal tightly.
7. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

1. Place the bottles in the cooler with ice and close the lid.
2. Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).

At the lab, store samples in a refrigerator at 4°C and process them within 24 hours.

1. +++END-IF+++
2. +++IF determine('Saltwater Water Quality','Saltwater','Total suspended solids','') === true +++

#### Total Suspended Solids

This section describes the procedures and methods for the field collection and preservation of the water samples to measure total suspended solids. Prior to sample collection, a label (including station location, replicate number, and date) will be taped to the outside of sample containers. The label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling collection device will be deployed.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 1 L sample container
* Cooler with ice
* Plastic electrical tape
* De-ionized water

For recording measurements:

* Sample collection form
* Field Data Form
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

Method (using a sample bottle):

1. Attach a label to a 1 L HDPE bottle and cover it with clear plastic tape.
2. Put on nitrile gloves.
3. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap.
4. Rinse the HDPE bottle three times with site water.
5. Hold the bottle near its base and plunge it (opening downward) below the water surface on the opposite side of the boat. Collect a water sample 0.1 m beneath the surface.
6. Recap the bottle carefully, remembering not to touch the inside.
7. Place the sample in a cooler on ice.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn):

1. Attach a label to a 1 L HDPE bottle and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the HDPE bottle with sample water.
6. Recap the bottle and seal tightly.
7. Place the sample in the cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

1. Place the bottles in a cooler with ice and close the lid.
2. Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).

At the lab, store samples in a refrigerator at 4°C and process within 24 hours.

1. +++END-IF+++
2. +++IF determine('Saltwater Water Quality','Saltwater','Water transparency (Secchi depth)','') === true +++

#### Water Column Transparency (Secchi Depth)

Water column transparency is measured using a Secchi disk. A Secchi disk is a 20 cm black and white disk suspended from a non-stretch line that is marked in 0.5 m intervals. A Secchi disk is used to measure the water column to nearest 0.1 m transparency at every station. If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the Secchi disk is deployed.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form and deploy the Secchi disk. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Measurements are made at prescribed depths as the Secchi disk is lowered and then again while it is retrieved, starting just below the surface, progressing down to 0.5 m from the bottom, and returning to just below the surface. Below are step-by-step procedures for measuring water column transparency.

Supply list for collecting samples:

* Secchi disk with rope marked at 0.5 m intervals
* Measuring tape

For recording measurements:

* Field Data Form
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

Method:

1. Slowly lower the Secchi disk until it is no longer visible. Record the depth where the marking on the line meets the water level. Interpolate between the 0.5 m markings on the rope to the nearest 0.1 m.
2. If the disk hits the bottom before disappearing, water column transparency depth is greater than the water depth.
3. Slowly raise the Secchi disk until it just becomes visible and record the depth. Interpolate between the 0.5 m markings on the rope to the nearest 0.1 m.
4. Repeat steps 1–3 two more times, recording both disappearance and reappearance depths each time.
5. Repeat the entire process if any one disappearance or reappearance measurement differs from the others by more than 0.5 m.

Record the data on the Field Data Form.

NOTE: For valid Secchi depth readings, no sunglasses, hats, or any other devices that shade the eyes may be used by the person who is observing the disappearance and reappearance depths. The Secchi depth is assessed from the shady side of the boat and can only be measured during daylight hours. One team member must make all three sets of Secchi measurements at a site, and it is desirable to have the same team member complete Secchi depth readings throughout the entire field season whenever possible.

1. +++END-IF+++
2. +++END-IF+++

+++IF determine('Saltwater Water Quality','Saltwater','Total Kjeldahl nitrogen','') === true || determine('Saltwater Water Quality','Saltwater','Total nitrogen','') === true || determine('Saltwater Water Quality','Saltwater','Ammonia-N','') === true || determine('Saltwater Water Quality','Saltwater','Nitrate-Nitrite-N','') === true || determine('Saltwater Water Quality','Saltwater','Total phosphorus','') === true || determine('Saltwater Water Quality','Saltwater','Orthophosphate','') === true +++

### B2.4 Nutrients

This section describes the procedures for the field collection and preservation of the water samples to be analyzed for nutrients including total nitrogen, nitrates, nitrites, ammonia, total phosphorus, and orthophosphates. Prior to sample collection, a label (including station location, replicate number, and date) will be taped to the outside of sample containers. The label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling collection device will be deployed.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the boat settles, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 1 L HDPE opaque bottles
* Cooler with ice
* De-ionized water
* Van Dorn or Niskin bottles (if using)
* Cooler with ice

For recording measurements:

* Sample collection form
* Field Data Forms
* Sample label with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)

Method (using a sample bottle):

1. Attach a label to a 1 L HDPE bottle(s) and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap.
4. Rinse the pre-labeled HDPE bottle three times with site water.
5. Hold the bottle near its base and plunge it (opening downward) below the water surface on the opposite side of the boat. Collect a water sample 0.1 m beneath the surface.
6. Recap the bottle carefully, remembering not to touch the inside.
7. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn):

1. Attach a label to a 1 L HDPE bottle(s) and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the 1 L HDPE bottle with sample water.
6. Recap the bottle and seal tightly.
7. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

1. Place the bottles in a cooler (on ice or water) and shut the lid.
2. Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
3. At the lab, store samples in a refrigerator at 4°C and process within 24 hours. If not analyzed within this time, the samples should be filtered and the filters frozen for future analysis (within 21 days of their collection).

If samples cannot be delivered to the laboratory within 6–8 hours following collection, acid preservation may be required. See “Method: Processing nutrient samples” below.

Processing nutrient samples:

Method

1. Draw dissolved inorganic nutrient sample water up from a transfer bottle (1 L opaque HDPE) using a 60-mL acid-washed syringe.
2. Push through an in-line Nuclepore 4.7 cm diameter 0.4 µm membrane filter and a 100 mL pre-labeled Whirl Pak.

Rinse the syringe with de-ionized water and triple-rinse it with site water between each sample. Also rinse the syringe with Milli-Q (de-ionized) between each station. Keep samples on ice until they are transferred to the lab; freeze them within 8 hours of collection.

1. +++END-IF+++

+++IF determine('Saltwater Water Quality','Saltwater','Chlorophyll-a','') === true +++

### B2.5 Chlorophyll *a*

This section describes the procedures and methods for the field collection and preservation of the water samples to measure chlorophyll *a*. Prior to sample collection, labels (including station location, replicate number, and date) will be taped to the outsides of sample containers. The label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling collection device will be deployed.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the boat settles, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 1 L opaque HDPE bottles
* De-ionized water
* Van Dorn or Niskin bottles (if using)
* Cooler with wet ice

The following items will also be needed for recording measurements:

* Sample collection form
* Field Data Form
* Sample label with pre-printed sample ID
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)
* Clear tape strips

Method (using a sample bottle)

1. Attach the completed label to the 1 L HDPE sample bottle(s) and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device (such as a Niskin, Van Dorn, or Kemmerer bottle) and the sample containers three times with water from the site.
4. Using either a water sampling device (such as a Niskin, Van Dorn, or Kemmerer bottle) or water pumping system, collect a water sample at 0.5 m below the surface (or mid-depth if station depth is less than 1.0 m).
5. Fill the HDPE bottle(s) with sample water.
6. Replace the cap(s) and seal tightly.
7. Place the sample in a cooler on ice at 4°C.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn):

1. Attach a label to a 1 L HDPE bottle(s) and cover it with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the HDPE bottle(s) with sample water.
6. Replace the cap and seal tightly.
7. Place sample in athe cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

* No chemical preservation in the field is needed.
* Place the bottles in a cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store samples in a refrigerator at 4°C and process them within 24 hours of the collection time.
* If not analyzed within this time, the samples will be filtered and the filters frozen for future analysis (within 21 days of their collection). See Method: Filtering Chlorphyll a Samples.

Filtering Chlorophyll a Samples

* Unless samples are taken to a laboratory for processing within 6 hours, the chlorophyll *a* sample must be processed and submitted to the laboratory as residue on a Whatman GF/F filter. Upon receipt of the filters, the laboratory extracts the pigment from the filter and quantifies it using fluorimetry. A portion of the filtrate produced from collecting the chlorophyll *a* sample is submitted to the laboratory and processed for dissolved nutrients.

Supply list for processing samples:

* Whatman GF/F 47 mm 0.7 μm filter
* 500 mL side-arm filter flask
* Filtration unit (blue base filter funnel, 250 mL unit)
* Rubber stopper and large filter funnel adapter vacuum pump (electric or hand)
* De-ionized water
* Nitrile gloves
* Forceps
* Graduated cylinder (250 mL)

For recording measurements:

* Sample collection form
* Chlorophyll *a* and dissolved nutrients sample labels
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)
* Clear tape strips

For sample collection and preservation:

* Centrifuge tube (50 mL, screw-top)
* Aluminum foil square
* HDPE bottle (250 mL, white)
* Cooler with dry ice
* Electrical tape
* Plastic bag (sandwich size)

Chlorophyll *a* samples should be processed in subdued light, out of direct sunlight.

1. In low light conditions, set up the filter apparatus with vacuum flask, filter holder, glass fiber filter, and filter funnel.
2. Using a clean graduated cylinder, measure a precise volume and record the amount on the Field Data Form.
3. Pour the measured sample into the clean filter funnel and filter with a vacuum pump a minimum of 500 mL of the sample until a visible quantity of green or greenish brown shown up on the filter. NOTE: If you don’t see more than a tinge, filter more sample. Filtering may significantly slow in the later stages as the filter plugs up with material.
4. Record the volume filtered to the nearest mL.
5. Remove the filter funnel, and carefully remove the filter from the filter holder using forceps. Fold the filter in half (green side in), and place in an air-drying box and cover.
6. Rinse all equipment (cylinder, filtering apparatus, and forceps) with distilled water before processing additional samples.

When all samples have been filtered, plug in the drying box. Air dry the sample filters for at least 45 minutes or until they are dry. Remove filters with forceps and place in aluminum foil. Label the aluminum foil; prepare it for delivery to the laboratory or freeze it.

1. +++END-IF+++
2. +++IF determine('Saltwater Water Quality','Saltwater','Enterococci','') === true +++

### B2.6 Enterococci

This section describes the procedures and methods for the field collection and preservation of the water samples to measure Enterococci. Prior to sample collection, a label (including station location, replicate number, and date) will be taped to the outside of sample containers. The label may be pre-printed and taped on using clear tape or written directly on write-on colored tape or a pre-printed adhesive label. Once the sampling crew is on station and coordinates have been verified, the sampling collection device will be deployed.

Enterococci levels tend to be highest in the morning prior to high levels of solar irradiation; therefore, these samples must be collected as early in the day and with as little water and sediment disturbance as possible. Regardless of when the enterococci samples are collected, must be filtered within 6 hours from collection.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* HDPE bottle (250 mL, clear, pre-sterilized)
* Sodium thiosulfate tablet
* De-ionized water
* Cooler with ice
* Van Dorn or Niskin bottles (if using)

The following items will also be needed for recording measurements:

* Sample collection form
* Field Data Form
* Sample labels pre-printed with sample ID
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels)
* Clear tape strips

Method: Sample Collection

1. Attach a label to the 250 mL sample bottle.
2. Put on nitrile gloves.
3. Lower the un-capped, inverted 250 mL sample bottle to a depth of 0.5 m below the water surface (or mid-depth if station depth is less than 1.0 m). Avoid surface scum, vegetation, and substrates. Point the mouth of the container away from the boat. Right the bottle and raise it through the water column, allowing it to fill completely.
4. Add the sodium thiosulfate tablet, cap, and shake the bottle 25 times.
5. Replace the cap and seal tightly.
6. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Method (using a sampling device such as a Niskin or Van Dorn bottle):

1. Attach a label to the 250 mL bottle and cover with clear plastic tape.
2. Put on nitrile gloves.
3. Rinse the sampling device and the sample containers three times with water from the site. Discard the water away from the sampling location if additional water is to be collected.
4. Deploy the sampling device to collect a water sample at 0.5 m below the surface and/or at 0.5 m off the bottom.
5. Fill the sample bottle with sample water.
6. Add the sodium thiosulfate tablet, cap, and shake the bottle 25 times.
7. Replace the cap and seal tightly.
8. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

* Add the sodium thiosulfate tablet, cap, and shake the bottle 25 times.
* Store the sample in a cooler on wet ice to chill (not freeze) for at least 15 minutes. Do not hold samples longer than 6 hours before filtration and freezing.
* Place the bottles in the cooler (on ice or water) and shut the lid.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* At the lab, store samples in a refrigerator at 4°C. Samples will need to be delivered to the laboratory within 6 hours of collection.

+++END-IF+++

+++IF determine('Saltwater Water Quality','Saltwater',' Microcystins','EPA 544') === true || determine('Saltwater Water Quality','Saltwater',' Microcystins','EPA 546') === true +++

### B2.7 Harmful Algal Blooms: Microcystins

The algal toxin (microcystin) sample is a grab sample taken from the site. The grab sample is collected using the 3 L beaker to fill two 500 mL bottles. A screening test is conducted in the field using dipsticks. If presence of microcystins is detected, and their concentration is higher than acceptable, the sample may be taken to the laboratory for further analysis.

If collecting samples using a boat, locate the station using a GPS. When close to the station, position the boat slightly up-current or upwind, with the side from which the sample is collected facing into the wind. (By setting up this way, the expected drift of the vessel will bring the vessel to the station coordinates when the sample is collected.) Deploy the anchor. Once the vessel has settled, record the coordinates on the Field Data Form. Record data pertaining to weather, sea state, and other ambient conditions about the site on the Field Data Form.

Supply list for collecting samples:

* Nitrile gloves
* 3 L Nalgene beaker
* HDPE bottle (500 mL white, round)
* Cooler with ice
* Algal toxin strip test kit for microcystins

For recording measurements:

* Sample collection form
* Field Data Form
* Sample labels with pre-printed sample ID
* Clear tape strips
* Pencils (for data forms)
* Fine-tipped indelible markers (for labels) (for labels)

Method: Algal Toxin Strip Test for Microcystin

1. Pour 1–2 mL of the sample from the HDPE bottle into the small bottle provided with the test kit.
2. Using the graduated pipette provided, transfer 1 mL of sample to the lysis vial containing the dried lysis reagent.
3. Cap the bottle and shake for 2 minutes. Let rest for 8 minutes.
4. Using the forceps provided, add one reagent paper to the lysis vial.
5. Cap and shake for 2 minutes. Let rest for 8 minutes.
6. Using the pipette provided, add seven drops of sample to the conical, flip-top tube containing the reagent.
7. Close the conical, fliptop tube and shake it for 30 seconds. Sample will turn purple.
8. Insert test strip into conical, fliptop tube with arrow pointing down (i.e., with the sample pad down). Incubate for 10 minutes.
9. Remove the test strip. Lay flat and allow to continue developing for 5 minutes.
10. Use the strip control and test lines to measure the approximate concentration of microcystins observed.
11. Replace the cap and seal tightly.
12. Place sample in a cooler on ice.

Record the collection data on the sample collection form.

Sample storage and handling:

* Place the 500 mL bottles in a cooler (on ice or water) and shut the lid. If a cooler is not available, place the 500 mL bottles in an opaque garbage bag and immerse them in the stream.
* Record the sample IDs on the sample collection form along with the pertinent site information (site name, ID, date, etc.).
* Freeze as soon as possible until delivered to the laboratory.

+++END-IF+++

## B3 Sample Handling and Custody

This section describes the process of sample handling and custody in the field, in the laboratory, and during transport, taking into account the nature of the samples, the maximum allowable holding time before extraction or analysis, and available shipping options and schedules.

Labels with the following information will be attached to sample containers:

* Sample number
* Site ID
* Time and date of collection
* Preservation requirements
* Name of sampler and organization

Samples for shipment will be prepared as follows:

* All samples will be appropriately preserved and packaged for transport.
* If obtainable samples are missing, the Project Manager will determine corrective action (e.g., reschedule a site visit or return to the site that same day to complete collection of the missing samples).
* All samples will be labeled and the labels checked for completeness, legibility, accuracy, and consistency.
* Labels and forms will be reviewed to ensure consistent sample ID information.
* Each sample container will be inspected to make sure there are no leaks and that all containers are properly sealed.

The Field Coordinator will complete the chain of custody form(s) for samples shipped to a laboratory. A copy of each tracking form will be made and retained by the team. The original form will be sent in the container with the sample. Copies of all tracking forms will be included in the coolers when samples are sent to the labs.

## B4 Analytical Methods

Field and laboratory analyses will be conducted according to the methods listed in the Table. This does not apply to in-situ parameters measured using sensors in the field. This includes temperature, pH, dissolved oxygen, salinity and turbidity).

Table B4.1. Methods of Detection for Analytes

|  |  |
| --- | --- |
| Parameter - Method | Typical MDL (mg/l or as stated) |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality' && param.mdl !== '') +++ |  |
| +++ **INS $**parameter.label+++ | +++ **INS $**parameter.mdl+++ |
| +++END-FOR parameter +++ |  |

## B5 Field and Data Quality Control

The monitoring project will include appropriate field and laboratory QC samples to assess general data quality issues, as well as specific data quality objectives. Field QC samples will be taken for 10% of all water quality samples taken per sampling event.

### B5.1 Field Duplicates

Duplicates will be taken side by side and simultaneously. Field duplicates are submitted to the laboratory along with all other samples. Field duplicates will be collected from 10% of the total samples to detect both natural variability in the environment and that cased by field sampling methods.

### B5.2 Field Blanks

Ambient field blanks will be taken at 10% of total samples to evaluate if any sample contamination may have occurred due to improper sample collection, atmospheric fallout, or other causes. A field blank will be created by filling a clean sample bottle with de-ionized or distilled water in the field during sampling activities, then treated the same as other samples taken from the field (i.e., labeled, stored on wet ice in a cooler). Field blanks are submitted to the laboratory along with all other samples and are used to detect any contaminants that may be introduced during sample collection, fixing, storage, analysis, and transport.

The QC protocols of the contracted laboratory shall be discussed with the laboratory personnel prior to sampling to ensure acceptability.

Table B5.1. Field Quality Assurance/Quality Control Summary

| Instrument/  Parameter | Accuracy Checks | | | Precision Checks | % Field QC Samples (blanks & field duplicates) | |
| --- | --- | --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality') +++ | |  |  | | |  |
| +++ **INS $**parameter.label+++ | | +++ **INS $**parameter.accuracyChecks+++ | +++ **INS $**parameter.precisionChecks+++ | | | +++ **INS $**parameter.percentFieldQC+++ |
| +++END-FOR parameter +++ | |  |  | | |  |

### B5.3 Quality Control Procedures: Field Operations

Field data quality is addressed, in part, by application and consistent performance of valid procedures documented in the standard operating procedures. Field crews will verify that all sample containers are uncontaminated and intact, and that all sample labels are legible and intact.

Before leaving the field, the crews will:

* Check sample labels to ensure that all written information is complete and legible.
* Place a strip of clear packing tape over the label, covering the label completely.
* Record the sample ID number assigned to the water chemistry sample on the sample collection form.
* Enter a flag code and provide comments on the Field Data Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity.
* Store the samples on wet ice in a cooler. Keep chlorophyll *a* filters frozen until shipping on wet ice.
* Recheck all forms and labels for completeness and legibility.

### B5.4 Field Quality Control: Multi-Parameter Units

For in situ measurements, each field instrument (e.g., multi-parameter unit) used by the crews must be calibrated, inspected prior to use, and operated according to manufacturer specifications. For instruments that are factory calibrated and checked, teams shall ensure that factory-certified diagnostics have been completed according to manufacturer specifications (preferably conducted immediately prior to the sampling season). sensors such as these do not require the daily calibration steps or the weekly diagnostic/QC solution checks.

Table B5.2. Field Quality Control: Summary, Multi-Parameter Unit

| Parameter | Check Description | Frequency | Acceptance Criteria | Corrective Actions |
| --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality')+++ |  |  |  |  |
| +++ **INS $**parameter.parameter+++ |  | +++ **INS $**parameter.frequency+++ |  | +++ **INS $**parameter.correctiveAction+++ |
| +++END-FOR parameter +++ |  |  |  |  |

Table B5.3. Data Validation Quality Control, Multi-Parameter Units

| Check Description | Frequency | Acceptance Criteria | | Corrective Actions |
| --- | --- | --- | --- | --- |
| Verify performance of temperature probe using wet ice. | Prior to initial sampling, daily thereafter | Functionality = ±0.50C | | See manufacturer’s directions. |
| Verify depth against markings on cable | Daily | ± 0.2 m | | Re-calibrate |
| pH - Internal electronic check if equipped; if not check against Quality Check Solution | At the beginning and end of each day | Alignment with instrument manufacturer’s specifications; or QCS measurement in range | | AM: Re-calibrate  PM: Flag day’s data. pH probe may need maintenance. |
| Check DO calibration in field against atmospheric standard | At the beginning and end of each day | ±1.0 mg/l | | AM: Re-calibrate  PM: Flag day’s data. Change membrane |
| Conductivity – internal electronic check if equipped; if not check against QCS | At the beginning and end of each day  Alignment with instrument manufacturer’s specifications | | AM: Re-calibrate  PM: Flag day’s data. Instrument may need repair. | |

### B5.5 Field Quality Control: Secchi Depth

No field calibration procedures are required for the Secchi disk. QC procedures include designating a specific crew member as the Secchi depth reader, taking all measurements from the shady side of the boat (unlike LICOR measurements, which are taken from the sunny side), and not wearing sunglasses or hats when taking Secchi readings.

### B5.6 Field Quality Control: Chlorophyll a and Nutrients

Field data quality is addressed, in part, by application and consistent performance of valid procedures documented in the standard operating procedures. That quality is enhanced by the training and experience of project staff and documentation of sampling activities. Field crews will verify that all sample containers are uncontaminated and intact, and that all sample labels are legible and intact. Before leaving the field, the crews will: check the label to ensure that all written information is complete and legible, place a strip of clear packing tape over the label, record the sample ID number assigned to the sample on the Sample Data Form, provide comments on the Sample Data Form if there are any problems in collecting the sample or if conditions occur that may affect sample integrity. Samples will be stored on wet ice in a cooler.

Table B5.4. Field Quality Control Activities: Chlorophyll *a*

| Quality Control Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Chlorophyll-a Containers and Preparation | Rinse collection bottles 3x with ambient water before collecting water samples. | Discard sample. Rinse bottle and refill |
| Holding Time | 24 hours | Qualify samples |
| Sample Storage | Chl a samples are shipped on wet ice | Qualify sample as suspect |
| Filtration | Use Whatman 0.7 μm GF/F filter. Filtration pressure should not exceed 3.4 psig to avoid rupture of fragile algal cells.  Rinse sample bottle for dissolved nutrient 3x with 10-20 mL of filtrate before collecting 250 mL of filtrate for analysis. | Discard and refilter |

Table B5.5. Sample Field Processing Quality Control Activities: Nutrients

|  |  |  |
| --- | --- | --- |
| Quality Control Activity | Description and Requirements | Corrective Action |
| Water Chemistry Container and Preparation | Rinse collection bottles 3xwith ambient water before collecting water samples. | Discard sample. Rinse bottle and refill. |
| Sample Storage | Store samples in darkness at 4°C.  Ship on wet ice within 24 hours of collection. | Qualify sample as suspect for all analyses. |

Table B5.6. Data Validation Quality Control: Chlorophyll a and Nutrients

| Activity or Procedure | Requirements and Corrective Action |
| --- | --- |
| Range checks, summary statistics, and/or exploratory data analysis | Current reporting errors or qualify as suspect of invalid |
| Review holding times | Qualify value for additional review |
| Review data from QA samples | Determine the impact and possible limitations on overall data usability |

### B5.7 Field Quality Control: Enterococci

Table B5.7. Field Quality Control Activities: Enterococci

|  |  |  |
| --- | --- | --- |
| Quality Control Activity | Description and Requirements | Corrective Action |
| Check integrity of sample containers and labels | Clean, intact containers and labels | Obtain replacement supplies |
| Sterility of sample containers | Sample collection bottle and filtering apparatus are sterile and must be unopened prior to sampling. Nitrile gloves must be worn during sampling and filtering | Discard sample and recollect in the field. |
| Sample Collection | Collect sample at the last transect to minimize holding time before filtering and freezing | Discard sample and recollect in the field. |
| Sample holding | Sample is held in a cooler on wet ice until filtering. | Discard sample and recollect in the field. |
| Field Processing | Sample is filtered within 6 hours of collection and filters are frozen on dry ice. | Discard sample and recollect in the field |
| Field Blanks | Field blanks must be filtered at 10% of sites. | Review blank data and flag sample data. |

Table B5.8. Data Validation Quality Control: Enterococci

| Check Description | Frequency | Acceptance Criteria | Corrective Action |
| --- | --- | --- | --- |
| Duplicate sampling | Duplicate composite samples collected at 10% of sites | Measurements should be within 10 percent | Review data for reasonableness; determine if acceptance criteria need to be modified |
| Field filter blanks | Field blanks filtered at 10% of sites | Measurements should be within 10 percent | Review data for reasonableness; determine if acceptance criteria need to be modified |

Temporal repeat samples are collected to estimate site measurement and index period variance. Repeat sampling provides data that can be used to evaluate the potential for the sampling design to estimate status and detect trends in the target site population.

During the field season, teams will revisit approximately 10% of the target sites. In order to ensure that sampling procedures are as comparable as possible from the first visit to the second visit, the same team who initially sampled the site also conducts the revisit. During site revisits, the full set of samples and in situ measurement parameters are collected. When sampling sites are identified as revisit sites, enterococci filter blanks must be collected during both the initial visit and the revisit. The filter blanks must be collected beforethe sample is filtered. See Section B3.2 for the procedure for collecting filter blanks.

### B5.8Field Quality Control: Microcystins

Table B5.9. Field Quality Control Activities: Microcystins

| QC Activity | Description and Requirements | Corrective Action |
| --- | --- | --- |
| Check integrity of sample containers and labels | Clean, intact containers and labels | Obtain replacement supplies |
| Holding time | Hold sample on wet ice and freeze immediately upon return to base. Keep frozen until shipping | Quality samples |
| Sample storage | Store samples in darkness and frozen (-20C)  Monitor temperature daily | Qualify samples as suspect |

Table B5.10. Data Validation Quality Control: Microcystins

|  |  |
| --- | --- |
| Activity or Procedure | Requirements and Corrective Action |
| Range checks, summary statistics, and/or exploratory data analysis | Current reporting errors or qualify as suspect of invalid |
| Review holding times | Qualify value for additional reviews |
| Review data from QA samples | Determine impact and possible limitations on overall data usability |

## B6 Instrument/Equipment Inspection and Testing

All equipment used to in-situ parameters or collect samples will undergo periodic maintenance and calibration verification performed by manufacturer’s representatives or service consultants. These procedures will be documented by date and the signature of the person performing the inspection. kept in good repair as per manufacturer’s recommendations to ensure proper function. Records of equipment inspection, maintenance, repair, and replacement will be kept in a logbook, along with standard operating procedures for instrument maintenance and calibration.

Table B6.1. Typical Instrument/Equipment Inspection and Testing Procedures

| Equipment | Inspection frequency | Type inspection | Maintenance, Corrective Action |
| --- | --- | --- | --- |
| Nutrient sample bottles | Before each use | Visual for integrity, cleanliness | Acid washed prior to use |
| Filtering apparatus | Before each use | Proper functioning, clean storage | Spare filters, syringe |
| Secchi disk, calibrated line | Before each use | Visual for integrity, cleanliness | Wipe tape. Spare disk, spare line. |
| Meters | Before each use | Battery life, membrane condition | Spare batteries, spare membranes |
| Sampling device | Before each use | Visual for integrity, cleanliness | Repair, replace if necessary |

## B7 Field Equipment Calibration

This section describes how continued quality performance of equipment and instruments is ensured. No analytical laboratory instruments are covered by this QAPP.

### B7.1 Pre-measurement Instrument Checks and Calibration

Field instruments shall be tested and calibrated prior to sampling. Equipment can be calibrated either prior to departure for the site or at the site. Site location will be verified using a GPS. Field crews will have access to backup instruments if any instruments fail the manufacturer performance tests or calibrations. Prior to departure, the following checks and calibrations shall be performed:

* If using a handheld GPS unit, turn on the GPS receiver and check the batteries. Replace batteries immediately if a battery warning is displayed. Boat-mounted GPS units run off of the boat electrical system.
* Test and calibrate the multi-parameter unit. Each field crew shall follow the manufacturer's calibration and maintenance procedures to calibrate multi-parameter sensors according to manufacturer specifications. Once each week, crews shall verify that the sensor is functioning properly by performing manufacturer recommended internal diagnostic readouts (e.g., pH millivolts, cell constants, and/or other diagnostic readings). Records of these checks will be saved in a logbook or other documentation. For units that do not have internal check capabilities, crews will need to verify that the sensor is measuring the parameters properly by measuring a commercially available QC solution with properties similar to the multi-parameter unit’s standard/confidence solution.
* Record pre-measurement calibration data on the instrument calibration form.

#### ￼The dissolved oxygen, pH, temperature and conductivity sensor functions of the multi-parameter or individual sensor units will be calibrated prior to departure to the sample site(s). A single calibration is sufficient for the day.

Table B7.1. Instrument Calibration Procedures

| Parameter | Instrument | Type of Inspection | Inspection and Calibration Frequency | Standard of Calibration Used | Corrective Action |
| --- | --- | --- | --- | --- | --- |
| +++FOR parameter IN parameters.filter((param) => param.monitoringCategory === 'Saltwater Water Quality') +++ |  |  |  |  |  |
| +++ INS $parameter.parameter+++ | +++ INS $parameter.instrument+++ |  | +++ INS $parameter.calibrationFrequency+++ | +++ INS $parameter.calibrationStandard+++ | +++ INS $parameter.correctiveAction+++ |
| +++END-FOR parameter +++ |  |  |  |  |  |

Notes:

* For instruments that are factory calibrated and checked, ensure that factory-certified diagnostics have been completed according to manufacturer specifications (preferably conducted immediately prior to the sampling season). sensors such as these do not require the daily calibration steps or the weekly diagnostic/QC solution checks.
* Once each week, verify that the sensor is functioning properly by performing manufacturer-recommended internal diagnostic checks. These are manufacturer- and model-specific, but typically involve accessing internal diagnostic readouts (e.g., pH millivolts, cell constants, and/or other diagnostic readings). Record results in a logbook or other documentation and save them.
* For sensors without internal check capabilities, check pH against a commercially available QC solution with properties similar to confidence solution of the instrument being used. Record the successful completion of the internal checks or the expected values and measured values of the QC solution.
* If using a commercially purchased pH QC solution for the weekly quality checks, follow the guidelines below:
* The pH QC solution containers should be labeled with expected values and preparation dates.
* The pH of the QC solution should approximate the pH expected at sampling sites.
* Bulk solutions should be replaced according to the manufacturer’s specifications or at any time if the crew suspects they have become contaminated.
* A commercially purchased primary conductivity/seawater standard can be used as the QC solution for weekly quality checks of conductivity/salinity. A secondary conductivity/seawater standard that is referenced against a certified standard may also be used.
* If a secondary standard is used, preparation and certification test procedures and results must be logged in a QA notebook and maintained by the state or contractor in-house QA personnel.
* The standard should be representative of the conditions expected in the field (~0.5–35 ppt for marine waters).
* The conductivity/seawater calibration standard and QC solution containers must be labeled with expected values and preparation dates.
* The standards should be replaced with fresh solutions at least every 3–4 days to void contamination.
* Bulk solutions should be replaced according to manufacturer’s specifications or if the crew suspects that they may have become contaminated.
* At least once per sampling season calibrate the temperature sensor against a National Institute of Standards and Technology–traceable thermometer.

### B7.2 Post-Measurement Calibration Check

#### Multi-Parameter Unit

After the in situmeasurements have been completed for the sampling day, a post-measurement calibration check of the multi-parameter unit must be performed. To do this, pH and conductivity of one of each of the respective calibration standards that were used earlier in the day to calibrate the instrument must be measured and values recorded. If significant drift is detected (as defined the manufacturer), the sensor may need service; data collected since the last successful calibration and post-measurement calibration check should be flagged. Discontinue use of any sensor that is not functioning properly.

### B7.3 Instrument/Equipment Inspection, Testing Procedures

Equipment maintenance will be conducted routinely. Records of equipment inspection, maintenance, repair. and replacement will be recorded in a logbook.

## B9 Data Acquisition Requirements

Secondary data (historical reports, maps, literature searches, and previously collected analytical data) may be used in the preparation of the sampling plan. These data may come from sources such as:

* Prior reports specific to the area
* Results of state agency or other water quality monitoring data
* Pertinent data collected by federal agencies, such as USGS bathymetry data and NOAA weather records
* Surveys completed in the embayment or embayment system of interest, including those identified through MassBays’ Inventory of Plans and Assessments (<https://www.mass.gov/service-details/massbays-inventory-of-plans-and-assessments>)

## B10 Data Management

Field crew shall record sampling data on field sheets, review them, sign, and submit to the Field Project Coordinator. The Field Project Coordinator will review the forms and confer with the crew on any required corrective action. Field crew will fill out the chain of custody form for forwarding the samples (processed or unprocessed) to the laboratory. Each person who handles or transports samples will also sign the custody form upon receipt of samples. Chain of custody forms will accompany samples to the lab and back to the Monitoring Program Coordinator by mail or pickup after each analysis run is completed.

Once laboratory analyses are complete, the laboratory personnel will email or mail laboratory results to the Monitoring Program Coordinator. The Monitoring Program Coordinator and/or Data Management Coordinator will enter raw field and lab data electronically. Data are then compared with field sheets for accuracy. The original data forms will be stored in the organization’s office. Electronic backups and copies of data forms will be made and stored.

Data QC steps will be taken at several stages. Documentation of data recording and handling, including all problems and corrective actions, shall be included in all preliminary and final reports (Corrective Action Reporting Form attached). See Section A9 for record handling and storage procedures.

+++END-IF+++

# Section C. Assessment and Oversight

## C1. Assessment and Response Actions

This section identifies the number, frequency, and type of planned assessment activities that will be performed to assure implementation of this QAPP. These activities will be overseen by the Project Manager.

### C1.1 Assessments

**Field Sampling Readiness Review**

A field survey plan will reference the specific field activities to be conducted and lists of equipment provided.

**Field Sampling Internal Audit**

The Project Manager in coordination with the Field Coordinator will be responsible for periodic internal audits to verify that field sampling procedures and measurements are properly followed. The internal field audit checklist will include examination of the following (See Table):

• Field sampling records

• Sample collection, handling, and packaging procedures

• Adherence to the Field SOPs and this QAPP

• QA procedures

• Chain-of-custody

• Sample documentation

Results of internal field audits will be documented in QA reports to the Project Manager (Section C2).

**Laboratory Audits**

System audits are performed as described in each laboratory’s QA manual for internal auditing. Laboratory audits may be conducted by the contracted laboratory’s QA/QC Manager at the project start up and then periodically as part of its analytical monitoring program. The laboratory audit checklist will review the following:

* QA organization and procedures
* Personnel training and qualifications
* Sample log-in procedures
* Sample storage facilities
* Analyst technique
* Adherence to laboratory SOPs and this QAPP
* Compliance with QA/QC objectives
* Instrument calibration and maintenance
* Facility security
* Waste management
* Data recording, reduction, review, reports, and archival
* Cleanliness and housekeeping

Preliminary results of the systems audit will be discussed with the laboratory management staff. A written report that summarizes audit findings and recommends corrective actions as relevant will be prepared and submitted to the Laboratory Director for response and to the Project Manager. The results of the audit, including resolution of any deficiencies, will be included in the QA reports, as described in Section C2.

**Performance Evaluation Sample Assessment [for Benthic invertebrate samples]**

Proficiency testing for infaunal taxonomic analyses is accomplished through regular communication and inter-calibration of infaunal samples among taxonomists.

**Data Audits**

Data will be audited under the direction of the QA Manager as part of the data validation process (Section D1). Raw data will be reviewed for completeness and proper documentation. Errors noted in the data audits will be communicated to analyses and laboratory management and corrected data will be verified. Audits of the data collection procedures at contracted laboratory will be the responsibilities of the laboratories. Each laboratory is fully responsible for the verification and validation of the data it submits. Data must be submitted in QAPP-prescribed formats only. While work is in progress, the contracted laboratory’s QA Manager or his/her designee will conduct an inspection to evaluate the laboratory data-production process. All data must be reviewed by the contracted laboratories’ QA Manager or designee prior to submission to the Project Manager.

### C1.2 Assessment Findings and Corrective Action Responses

All technical personnel share responsibilities for identifying and resolving problems encountered in the routine performance of their duties. Issues that affect the schedule, cost, or performance of project tasks will be reported to the Project Manager. The Project Manager will be accountable for overall implementation of the Project. The Project Manager will be responsible for identifying and resolving problems that (1) have not been addressed in a timely manner or successfully at a lower level, (2) influence multiple components of the projects, or (3) require consultation with contracted laboratories. The Project Manager will be responsible for evaluation the overall impact of the problem on the project and for developing and implementing corrective actions. The Project Manager will also identify and resolve problems that may necessitate changes to this QAPP. Problems identified by the Field Coordinator and the QA Manager will be reported to the Project Manager and corrected as described in Section C2.

Table C1 Example of Internal Field Audit Checklist

The QA Manager will generate and/or review all corrective actions required during the project and monitor their effectiveness in meeting project quality objectives. Corrective action is the process of identifying, recommending, approving, and implementing measures to counter unacceptable procedures or out-of-limit QC performance that can affect data quality. Corrective action can occur during field activities, laboratory analyses, data validation and assessment. All corrective action proposed and implemented should be documented in the QA reports to the Project Manager. A copy of the QA/QC Corrective Action Log will be provided as described in Section C2.

**Field Corrective Action**

Corrective action in the field may be needed when the sample frequency is changed (i.e., more/fewer samples, sample locations other than those specified in the Study Plan), or when sampling procedures and/or field analytical procedures require modification due to unexpected conditions. The survey crew may identify the need for corrective action. The Project Manager and QA Manager will approve the corrective measure and ensure that the survey crew implements the corrective action.

Corrective action resulting from internal field audits will be implemented immediately if data may be adversely affected due to unapproved or improper use of approved methods. A corrective action issue which directly impacts the project DQOs will be reported to the Project Manager. Corrective action will be documented in QA reports to project management (Section C2). Corrective actions will be implemented and documented as follows:

* A description of the circumstances that initiated the corrective action
* The action taken in response
* The final resolution
* Any necessary approvals
* Effectiveness of corrective action

**Laboratory Corrective Action**

Corrective action in the laboratory is specified in laboratory SOPs and may occur prior to, during, and after initial analyses. Following consultation with laboratory analysts and supervisory personnel, it may be necessary for the QA Manager to approve the implementation of a corrective action. If the problem makes it impossible to achieve project objectives, the laboratory manager will be notified, who will in turn notify the Project Manager.

Corrective actions will be performed prior to release of the data from the contracted laboratory. The corrective action will be documented in both the laboratory’s corrective action files, and in the data report generated by the laboratory. If the corrective action does not rectify the situation, the laboratory will contact the Project Manager, who will determine the action to be taken.

**Corrective Action during Data Validation and Data Assessment**

The need for corrective action may be identified during either data validation or data assessment. Potential types of corrective action may include re-sampling by the survey crew or reanalysis of samples by the laboratory. These actions are dependent upon the ability to mobilize the survey crew and whether the data to be collected are necessary to meet the required QA objectives. If the data validator or data assessor identifies a corrective action situation that impacts the achievement of the project objectives, the Project Manager will be notified.

## C2. Reports

Data that have passed preliminary QC analysis (Section B5) will be uploaded to WQX and shared with interested audience. Any data uploaded or released will be accompanied by the caveat that they are for review purposes only and subject to correction after completion of a full data review at the end of the sampling season.

The Project Manager will prepare a final report which will be shared with the QAPP distribution list. The final report will include tables and graphs developed for initial data distribution efforts and will describe the program goals, methods, quality control results, data interpretation, and recommendations and include:

• Raw data

• QC data

* Secondary data used

• Associated metadata

• Questionable data flagged

• Preliminary or final report

• Other

# Section D. Data Review and Usability

## D1. Data Review and Validation

A review protocol is developed to ensure that data validation and verification is conducted in an objective and consistent manner. The review will include the required number, frequency and types of assessments (peer reviews, management systems reviews, technical systems audits, performance evaluations, and data quality reviews), and names of staff responsible for this task.

**Field Data**

The field data verification includes verification of sampling design, sample collection procedures, and sample handling. Field data will be reviewed regularly by the Project Manager to ensure that the records are complete, accurate, and legible and to verify that the sampling procedures are in accordance with the protocols specified in the QAPP (refer to Section D2.1 for the specific elements reviewed).

**Laboratory Data**

Prior to the release of any data from a contracted laboratory, the data will be reviewed and approved by laboratory personnel. The review will consist of a tiered approach (Section D2.2) that will include reviews by the person performing the work, by a qualified peer, and by supervisory and/or QA personnel.

**Data Management**

The review process will include verification of manually entered data and QC checks run in a software application prior to submitting the data to WQX. Detailed descriptions of these processes are included in Sections B10 and D2.

## D2. Verification and Valuation Methods

Data verification methods will ensure that the reported results reflect what was actually done and document that the data fulfill applicable requirements. Validation will further identify and evaluate the impact of any technical non-compliance or quality control non-conformances on the complete data set.

**Field Data**

Field records will be reviewed by the Project Manager to ensure that:

* Logbooks and standardized forms have been filled out completely and that the information recorded accurately reflects the activities that were performed
* Records are legible and in accordance with good recordkeeping practices, i.e., entries are signed and dated, data are not obliterated, changes are initialed, dated, and explained
* Equipment calibration, sample collection, handling, preservation, storage, and shipping procedures were conducted in accordance with the protocols described in this QAPP, and that any deviations were documented and approved.
* DQIs are calculated and results compared with DQOs for review by the QA Manager; and data compares well to historic data or checking its “reasonableness.”

**Laboratory Data**

As a part of data validation, contracted laboratories will ensure that:

* The QC checks specified in Sections A7 and B5 were conducted and met the acceptance criteria
* All data that are hand-entered (i.e., typed) will be 100% validated prior to use in calculations or submission to the Project Manager
* All manual calculations will be performed by a second staff member to verify that calculations are accurate and appropriate
* Calculations performed by software will be independently verified at a frequency sufficient to ensure that the formulas are correct, appropriate, and consistent, and that calculations are accurately reported

Once data have been generated and compiled in the laboratory, laboratory personnel will review the data to identify and make professional judgments about any suspicious values. All suspect data will be flagged and reported. These data may not be used in calculations or data summaries without the review and approval of the appropriate senior staff. No data measurements will be eliminated from the reported data or database and data gaps will never be filled with other existing data. The loss of any samples during shipment or analysis will be noted in the database.

**Data Management**

Laboratory data will be reviewed by the Project Manager prior to the electronic submission to WQX. Data review may include methods such as plots, logical checks, and range checks to identify suspect values. Routine system back-ups are performed daily. Data provided electronically to facilitate data handling will be verified against the hard copy data. Detailed description of data management and review is provided in section B10 of this QAPP.

**Project Deliverables**

Upon completion of the verification/validation process, a dataset packet will be prepared for submittal to WQX. The data will be in the format prescribed for submission to WQX. This documentation will include the following elements as listed in Section A9.4.

* Cover letter that includes a description of any problems
* List of problems encountered, and corrective action taken
* List of samples/images planned versus collected, or measurements planned versus reported
* Quality Assurance Statement including a checklist of QA actions, and notes on deviations and corrective actions
* Table(s) of data submitted

## D3. Reconciliation with User Requirements

This section describes how the verified/validated project data will reconcile with the project DQOs, how data quality issues will be addressed, and how limitations on the use of the data will be reported and handled. To meet these DQOs, a combination of qualitative evaluations and statistical procedures will be used to check the quality of the data. These procedures will be used by the laboratory generating the data, and by the Project Manager or a designee.

The data generated must meet the project DQOs defined in Section A7 of this QAPP. The primary objectives for assessing the usability of the data are to ensure that (1) data denote conditions and habitat quality in the area being studied, (2) all datasets are complete and defensible, and (3) data are of the quality needed to meet the overall objectives of the program.

### D3.1 Comparison to Measurement Criteria

**Accuracy and Precision Assessment**

The accuracy and precision of the data generated during this project will be assessed by comparison to the DQOs specified in Section A7. Data that fail to meet the data quality criteria may necessitate sample reprocessing, analysis of archival material, sample recollection, or flagging of the data, depending on the magnitude of the nonconformance, logistical constraints, schedule, and cost.

**Completeness Assessment**

Completeness is the ratio of the number of valid sample results to the total number of results planned for collection. The overall completeness goal for the monitoring program is 100% of planned samples to be collected and analyzed. The Project Manager will assess the completeness of the overall data generation against the project goals. Following completion of the sampling, analysis, and data review, the percent completeness will be calculated and compared to the project objectives stated in Section A7.2 using the following equation.

If this goal is not met, data gaps will require evaluation to determine the effect on the intended use of the data. Sample re-analysis, analysis of archived material, and/or re-collection of the sample may be appropriate depending on criticalness of the missing data, logistical constraints, cost, and schedule.

**Representativeness**

Representativeness expresses the degree to which data accurately and precisely denote a characteristic of a population, parameter variations at a sampling point, a process condition, or an environmental condition within a defined spatial and/or temporal boundary. Representativeness of the field data will be assessed by verifying that the sampling program was implemented as proposed and that proper sampling techniques were used. The assessment of representativeness in the laboratory will consist of verifying that the proper analytical procedures and appropriate methods were used.

### D3.2 Overall Assessment of Environmental Data

Data assessment will involve an evaluation to determine if the data collected are of the appropriate quality, quantity, and representativeness for the purposes required by project as well as for submission to WQX. This evaluation will be performed by the Program Manager in concert with other users of the data. Data generated in association with QC results that meet these objectives will be considered usable. Data that do not meet the objectives and/or the data validation criteria might still be usable. This assessment may require various statistical procedures to establish outliers, correlations between data sets, adequate sampling location coverage, etc., in order to assess the effect of qualification or rejection of data. The effect of the qualification of data or loss of data deemed unacceptable for use, for whatever reason, will be discussed and decisions made on corrective action for potential data gaps.

# Forms

[e.g., program specific data collection forms]

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