+++ IF 4 < 3 +++

**Color key for this file**

* Instructions to be placed into the application
* Tips to be placed into the application
* Developer code embedded in this document (do NOT touch)
* Important information that will be turned into Developer code
* Do not touch the developer code that looks like “**+++INS `${title}`+++**”
* Must be provided before the QAPP is considered complete and ready for review.
* Not sure what to do with

+++ END-IF +++

**Quality Assurance Project Plan**

**for**

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

**Prepared by**

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

**Approvals Signature** (required prior to project start):

+++FOR person IN projectOrganization +++

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

\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_ Date: \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

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

+++END-IF+++

+++END-FOR person+++

**Table of Contents**

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[A5.2 Monitoring History and Status 19](#_Toc12002390)

**SECTION A**

**A1 TITLE PAGE**

**A2 TABLE OF CONTENTS**

**A3 DISTRIBUTION LIST**

+++ IF 4 < 3 +++

Instructions: List the individuals and their organizations that need copies of the approved QAPP, including all persons responsible for implementation.

+++ END-IF +++

+++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**

*+++ IF 4 < 3 +++*

Instructions: List the individuals or organizations participating in the project, their specific roles and responsibilities. Provide a concise organization chart showing the relationships and lines of communication among project participants. Table 4.1 provides examples of relevant roles. It is important however that the (Program QA Officer’s) role is held by a specific person and separate from the roles held by data generators and data managers. Identify any subcontractors’ relationships relevant to laboratory work and data operations.

*+++ END-IF +++*

Table 4.1 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**

*+++ IF 4 < 3 +++*

Clearly describe the specific environmental problem(s), question(s) or threat(s) to be addressed by the project. Explain why the work needs to be done, identifying the reasons for conducting the work and/or collecting information relating to the project. Provide relevant historical information, previous studies, and data that may have been collected. Identify the data gaps that this project will fill. Describe the monitoring objectives of the project.

*+++ END-IF +++*

**A6.1 Problem Definition**

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

[CHECKLIST PROVIDED BY THE APP]

The monitoring objectives of this project include *(check all that apply)*:

* Provide quality-controlled data that assesses the condition of water quality and ecosystem health in freshwater rivers and streams
* Provide quality-controlled data that support the assessment and restoration of watersheds and critical habitats
* Water body/watershed health assessment
* Impact assessment
* Source identification or hot spot monitoring
* introduced species assessments
* Public education and outreach
* Local infrastructure improvements
* Other (specify) \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

**A6.2 Problem Background**

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

**A6 PROJECT DESCRIPTION AND TIMELINE**

**A6.1 Project Description**

*+++ IF 4 < 3 +++*

Describe how the project addresses the problem(s) or will answer environmental question(s) and links data results with possible actions. Provide a general summary of the project, including information on data to be collected, the technologies or methods used to collect data and the decisions you plan to make with the data to support your program objectives. Include information on project objectives and data users. Provide a description and map of the project site or study area, and sampling locations and how they were selected. Identify the time period for data collection.

*+++ END-IF +++*

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

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

*+++ IF 4 < 3 +++*

Include map(s) of area with sampling site.

*+++ END-IF +++*

[Specify format and size (for end user) that are compatible with the APP].

A6.3 Anticipated Schedule

*+++ IF 4 < 3 +++*

List all major project implementation and completion activities and mark associated dates with an X. Add additional project components and deliverables as necessary. This represents a typical revolving calendar. Some tasks may continue into the following year (e.g. specimen identification, data interpretation and reporting).

*+++ END-IF +++*

Table 6.1 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 +++* |  |  |  |  |  |  |  |  |  |  |  |  |

**A7 FRESHWATER / WATERQUALITY DATA QUALITY OBJECTIVES**

*+++ IF 4 < 3 +++*

Provide a qualitative or quantitative DQO statement for the project.

*+++ END-IF +++*

[SPACE FOR FREE TEXT]

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.

**[Can these show up as TIPS linked to the bold text above?]**

**Acc**uracy

While training and audits help to ensure measurement accuracy and precision, quantitative measures of accuracy for water quality monitoring will be estimated using laboratory QC data (blank results, fortified matrix results, known QC samples, etc.).

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

**Precision**

Duplicate measurements for at least 10% of samples per parameter/per sampling “event” will be taken where applicable. Sampling sites have been selected to be representative of the waterbody (or in the case of hotspot monitoring, of the pollution source of interest). Sample collection timing and frequency was designed to capture data that are representative of target conditions (e.g. a range of water levels, weather, seasons, etc.).

Precision of the multi-probe measurements will be determined by taking duplicate (via a second placement of the unit) readings at the same station location. Multi-probe precision objectives range from 5-10 % RPD depending on the parameter.

**Representativeness**

Sampling locations and survey times have been selected to ensure that the samples taken represent typical field conditions at the time and location of sampling, and not anomalies due to uncommon effects, except in the case of stations chosen to evaluate site-specific impacts (or “hot spots”) which dictate the representativeness of distinct conditions. Other factors, such as seasonality and weather conditions, will be recorded and considered by data users when evaluating whether the resulting data are representative of (e.g., wet weather water quality).

**Comparability**

Standardized sampling and analytical methods, units of reporting, and site selection procedures will ensure comparability of data with others using those same methods. Care will be taken to document sample time and date of collection, sample storage and transfer, as well as laboratories and identification specialists used so that future surveys can produce comparable data by following similar procedures. Efforts to enhance data comparability are made where possible and appropriate, for example reviewing existing data and methods used to collect historical data have been reviewed and taken into account in the sampling design.

**Completeness**

Completeness is expressed as a percentage of the number of valid measurements that should have been collected*.* For [Project name], 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 will be produced.

**Typical DQI 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 decisionmaking 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 |

**Detection Limits**

Method Detection Limits (MDL) are calculated in the laboratory. (Detection limits in the traditional sense do not apply to some measurements such as pH and temperature that have essentially continuous scales). Multiplication factors are typically applied to MDL values by labs to express Reporting Limits (RL) which define a level above which there is greater confidence in reported values. Where low-level results are needed, if possible the laboratory may report results down to the MDL value with qualification as appropriate (rather than “< RDL”).

*+++ IF 4 < 3 +++*

Instructions: Provide a table that lists the Data Quality Objectives (DQOs) Details of how these criteria are met for each component of the [monitoring program] are presented in Section B5.

*+++ END-IF +++*

Table 7.1 Data Quality Objectives for Freshwater water quality monitoring

| **Parameter** | **Units** | **Accuracy 1** | **Overall Precision 2**  **(RPD)** | **Approx. Expected Range3** |
| --- | --- | --- | --- | --- |
| Station depth | meters | +/- 0.1 meter | 10% | 0-15 meters |
| Temperature | Celsius (C) degrees | 0.15 | +/- 0.1 | 0-35 |
| pH | Std. Units | +/- 0.3 | +/- 0.1 or < 10% (between field duplicate samples or readings) | 4-10 |
| Dissolved Oxygen | mg/l | +/- 0.2 | +/- 0.2 or < 20% (between field duplicate samples or readings) | 0-14 |
| % Oxygen Saturation | % | 2% | 5% RPD | 0.2-110 |
| Conductivity | μS/cm | 1% of range | 5% | 10–1000 for fresh |
| Turbidity | NTU | 1% of full scale (0-10)  5% of full scale (0-100) | + 0.5 NTU < 1 NTU or 20% RPD if >1 NTU | 0-200 |
| Total Nitrogen (TN) | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | 0.02 or 25% | 0-2 |
| Ammonia (NH3-N) | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | 0.01 or 20% | 0-0.5 |
| Nitrate-Nitrite as N  (NO3-NO2-N) | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | 0.02 or 25% | 0-1 |
| Total Phosphorus (TP) | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | <50 ppb, 5ppb  >50 ppb, 10% RPD | 0-0.15 |
| Orthophosphates | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | <50 ppb, 5ppb  >50 ppb, 10% RPD | 0-0.15 |
| Total Suspended Solids | mg/l | 80% - 120% recovery for QC std. and lab fortified matrix (LFM) | 1.5 or 40% RDP | 0-100 |
| Chloride | mg/l | 90% - 110% recovery for QC std. and lab fortified matrix (LFM) | 20% | 0-100 |
| Chlorophyll a | μg/L | 75-125% for QC std. | 2.0 or 20% RPD | 0-100 |
| Total Microcystins  (field test) | μg/L | 0.2 (est.) | 20% | 1-10 |
| *E. coli.* | CFU/100 ml *or* MPN/100 ml | Blanks and negatives show no colonies, ***positives*** show colonies | For log10 transformed field duplicate data:  < 30% RPD (< 50)  < 20% (50-500)  < 10 % RPD (500-5000)  < 5% (>5000 CFUs) | 0-5000 |
| Precipitation | inches (rain gage) | +/- 0.1 inch (in general) | < 20% (between two different gages for the same event) | 0-3 inches per event |
| Location by coordinates (GPS) | degrees and decimal minutes (NAD 1983) | +/- 20 feet with Wide Area Augmentation System (WAAS) enabled | Repeated readings, record during maximum satellite coverage | NA |

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 Project QA/QC Officer will determine how the resulting dataset compares with the monitoring program’s data quality objectives (DQOs). This review will include, for each parameter, calculation of the following:

* Completeness goals: overall % of samples passing QC tests vs. number proposed in Section A7
* % 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 [Monitoring Program Coordinator and/or QA Officer] will evaluate the overall attainment of DQOs and determine what limitations to place on the use of the data, or if a revision of the DQOs is allowable.

**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.

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.

**A7.1 Sample Collection Data Quality Objectives (DQOs)** *–* ***if all parameters selected***

***Macrofaunal sampling DQOs***

The DQOs for the of benthic infauna are that 1) transects will be determined appropriately for the stream size and morphology, and all sample sites will be assessed, (2) at least 100 organisms will be collected per sample site.

***Benthic algal biomass sampling DQOs***

DQOs for assessment of benthic algal biomass and algal community are that (1) transects will be determined appropriately for the stream size and morphology, and all sample sites will be assessed, (2) at least 35 observations will be made with each sample site, (3) at least 75% of taxa will be identified, 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).

***Stream characteristics assessment DQOs***

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

**Accuracy**: Each data entry will be checked to the original field sheet 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:

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).

For physical habitat assessment, final conclusions are potentially subject to variability among investigators. This limitation will be minimized by (1) ensuring that each investigator is appropriately trained in the evaluation technique and (2) conducting periodic crosschecks

among investigators to promote consistency. 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.

**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**: 100% 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.

**A7.2 Sample Analysis Data Quality Objectives (DQOs)** *–* ***for macrofaunal analysis only***

***Macrofaunal analysis DQGs***

The DQGs 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).

**A8 TRAINING REQUIREMENTS**

*+++ IF 4 < 3 +++*

List any required training that an individual involved with the project would need.

*+++ END-IF +++*

Instruction in 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 (Table 8.1).

All members of the project team are required to attend training/workshops appropriate to the type of monitoring they will conduct. The Monitoring Program Coordinator 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 Monitoring Program Coordinator 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.

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

Table 8.1 Project Specific Training

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Training Type/Description** | **Trainer(s)** | **Date(s)** | **Personnel/Group to be Trained** | **Location of Training Records** |
| Field safety |  |  |  |  |
| Laboratory safety |  |  |  |  |
| Sample collection |  |  |  |  |
| Filling out field sheets |  |  |  |  |
| Data entry and database management |  |  |  |  |
| Recordkeeping and documentation |  |  |  |  |
| Report writing |  |  |  |  |
| Other: (*specify)* |  |  |  |  |
| Other: (*specify)* |  |  |  |  |

**A9 DOCUMENTATION AND RECORDS**

*+++ IF 4 < 3 +++*

Briefly describe the documents, forms and records utilized and procedures for handling and storing records. Using Table 9.1 describe the record handling procedures by answering the following questions:

* Document and record-keeping: How will you make sure everyone has the same approved version of the QAPP?
* Data generation: What documents and records will be produced during and for your project? Three basic areas include field data, laboratory data, and data assessment.
* Data report package: What specific records and/or documents will be included with the final data report? Data from literature searches and/or databases? Field logs? Audit reports? Other?
* Reporting format: are there hard copy and/or electronic reporting formats? If so, what are they?
* Storage: do you have requirements for storing the records and documentation? If so, what are they?

*+++ END-IF +++*

Table 9.1 Table describing record handling procedures

|  |  |
| --- | --- |
| **Activity** |  |
| Document and record-keeping |  |
| Data generation |  |
| Data report package |  |
| Reporting format |  |
| Storage |  |

Table 9.2 Project-specific Datasheets, Labels, Laboratory, and Voucher Forms

|  |  |  |  |
| --- | --- | --- | --- |
| **Document Type** | **Form Name** | **Description** | **Relevant to which QAPP** |
| Field data sheets |  | Completed on site at the time of sampling. | All |
| Site verification form |  | Completed as part of site assessment verification visit | All |
| Equipment Custody Form |  | Lists all equipment provided to volunteers. | All |
| Photo log |  | Completed on site at the time of sampling | Fw benthic; marine benthic |
| Flow velocity estimation |  | Completed on site at the time of sampling | Fw benthic |
| Algal biomass field sheet |  | Completed on site at the time of sampling | Fw benthic |
| Algal biomass reference sheet |  | Preparatory material before field visit | Fw benthic |
| Kick sample collection filed sheet |  | Completed on site at the time of sampling | Fw benthic |
| Rock basket field sheet |  | Completed on site at the time of sampling | Fw benthic |
| Invert sorting sheet |  |  | Fw benthic |
| Habitat assessment field sheet |  | Completed on site at the time of sampling | Fw benthic |
| Laboratory Data Sheets |  | These forms will include information on the lab SOPs, data analysis, QA/QC and results. | All |
| Chain-of-Custody Forms |  | These will accompany samples from collection sites to labs. | All |
| Sample labels |  | These will be placed on all sample containers and will include the site name, date, time, location, type of sample, and crew name. | All |
| Training and Evaluation Form |  | These will include information on volunteer training | All |
| Instrument Log |  | This will include information on maintenance, calibration and testing on equipment | All |
| Other |  |  |  |

**SECTION B**

Add a Fresh water / Water quality IF statement here

**B. FRESH WATER / WATER QUALITY DATA GENERATION AND ACQUISITION**

B1. Sampling Design

*+++ IF 4 < 3 +++*

Describe the overall design of the project data collection activities. Provide the rationale for the sampling design, selection of sampling locations, and data collection activities and methods, including the types of technologies employed. Include location-specific information (e.g. GPS location) or maps of the data collection locations. Provide a narrative statement in response to these questions:

* **Sampling design**: Are you planning a descriptive, statistical, or a targeted approach?
* **Locations:** At what locations are you sampling? Provide a site map.
* **Methods**: What are your data collection activities? Are these grab samples or transects? Are you collecting data using in situ electronic instruments?
* **Frequency**: How often are you taking samples?

**Quality Control**: What types of field QC samples (e.g. field blanks, replicates, co-located samples) will you collect?

*+++ END-IF +++*

PUT SOME TYPE OF SAMPLING DESIGN INFORMATION HERE

**[TIP linked to “Sampling Design” section above]**

**Sampling Design Considerations.** Typical sampling design principles for watershed/waterbody health assessments, impact assessments, habitat assessments and introduced species assessments are described in Guidance Document XX. These are further broken into subcategories for river, lake, and wetland monitoring as appropriate. When describing project-specific sampling processes, these procedural considerations shall be followed or modified to meet specific monitoring objectives.

Instructions: Include a map and detailed descriptions of the sampling locations. Photographs and GPS coordinates of sampling sites are also recommended. Provide a legend, scale, and compass direction.

Instructions: Provide the following information in the table(s) below for discrete samples.

* State the parameters that will be measured/sampled.
* Provide the number of sampling locations.
* State if multiple samples will be collected at each location, such as sampling at different depths or taking repeated measurements over a given amount of time (i.e. once/ quarter).
* State the number of quality control (QC) samples that will be collected.
* State the specific methods or SOPs that will be used. Attach as appendices.
* State the rationale for these samples, e.g. Why the data will be collected at the particular location, frequency and time. Use a short reference to the objective, for example, some stations might be considered “reference” stations, while others may be considered “impacted”.

Table 1.2 Data Collection Methods

| Parameter | 3Number of sample locations | **1Rationale for number of samples** | **2Site location rationale** | **4Frequency** | **Number/type of QC samples7** |
| --- | --- | --- | --- | --- | --- |
| Temperature |  |  |  |  |  |
| pH |  |  |  |  |  |
| Dissolved Oxygen |  |  |  |  |  |
| Conductivity |  |  |  |  |  |
| Turbidity |  |  |  |  |  |
| Chlorophyll-a |  |  |  |  |  |
| Total Nitrogen (TN) |  |  |  |  |  |
| Ammonium-N |  |  |  |  |  |
| NO3-NO2-N |  |  |  |  |  |
| Total Phosphorus (TP) |  |  |  |  |  |
| Orthophosphate |  |  |  |  |  |
| Total Suspended Solids |  |  |  |  |  |
| Chlorides |  |  |  |  |  |
| Microcystins |  |  |  |  |  |
| E. coli bacteria5,6 |  |  |  |  |  |
| Stream flow characterization | Use source indicated in freshwater benthic QAPP | | | | |

**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): \_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_\_

3At least one each for selected reach or tributary [TIPS]

4At least monthly; minimum three “dry” weather surveys; pre-dawn or early mornings DO especially useful [TIPS]

5 Number of sample locations: If measuring bacteria for known source, measure at least at two sites (one just upstream and one just downstream of impact/source). If source tracking, numerous samples might be needed to identify the likely source(s). [TIPS]

6 Frequency: If source is known, minimum of 3 times each site, including wet and dry weather. If source tracking, conduct “as needed” to locate source.

7 Number of field QC checks may include field duplicates (10%) and Field blanks (10%). Field blanks only apply to grab samples. Multi-meter measurements for temperature, pH, DO, conductivity and turbidity are replicated at a sampling site using all available instruments or at 10% of the sites by taking multiple readings with the same instrument if only one instrument is being used. [TIPS}

B2. SAMPLING METHODS: SAMPLE COLLECTION AND STORAGE

+++ IF 4 < 3 +++

Instructions: This section describes the procedures to be applied to collect field samples and identifies the sampling methods, sample preservation requirements, decontamination procedures, and materials needed. Select a parameter to be measured. If more than one option for measurement of the parameter is available, select from the list. Each parameter will include the following information:

* List of field equipment, materials, and supplies
* Equipment preparation requirements
* Instructions for sample collection and any procedures to homogenize, filter, and preserve the samples in the field
* Maximum holding times to sample extraction and /or analysis

+++ END-IF +++

Table 2.1 Equipment preparation, sample processing and storage requirements

|  |  |  |  |  |  |
| --- | --- | --- | --- | --- | --- |
| **Parameter** | **Sample collection method** | **Sample Container** | **Sample Volume** | **Sample Preservation** | **Maximum Holding Time** |
| Temperature | In situ |  |  |  |  |
| pH | In situ |  |  |  |  |
| Dissolved oxygen | In situ |  |  |  |  |
| Dissolved oxygen (Winkler titration) | Grab | See kit instructions | See kit instructions |  |  |
| Conductivity | In situ |  |  |  |  |
| Turbidity | Grab | 500 ml HDPE  Pre-cleaned | 500 ml | 40C in dark | 48 hrs |
| Chlorophyll a | Grab | 500 ml opaque brown HDPE  Pre-cleaned | 500 ml | 40C in dark | 24 hrs (filter)/21 d if frozen |
| Total Nitrogen (TN) | Grab | Whirlpak bag or  HDPE bottles  Pre-cleaned | 500 ml | 40C in dark | 28 d |
| Nitrate-Nitrite-N | Grab | Whirlpak bag or HDPE bottles  Pre-cleaned | 500 ml | 40C in dark | 28 d |
| Ammonium-N | Grab | Whirlpak bag or HDPE bottles  Pre-cleaned | 500 ml | H2SO4 to pH < 2.0, 40C in dark | 28 d |
| Total Phosphorus (TP) | Grab | Whirlpak bag or HDPE bottles  Pre-cleaned | 500 ml | H2SO4 to pH < 2.0, 40C in dark | 28 d |
| Orthophosphates | Grab | Whirlpak bag  HDPE bottles | 500 ml | 40C in dark;  filter preferred | 48 hrs |
| Chloride | Grab | 500 ml HDPE Pre-cleaned\* | 500 ml | 40C in dark | 28 d |
| Total suspended solids | Grab | 500 ml HDPE  Pre-cleaned | 500 ml | 40C in dark | 7 d |
| Microcystins | Grab | 500 ml opaque brown HDPE  Pre-cleaned | 500 ml | 40C in dark |  |
| E. coli | Grab | Whirlpak bag  HDPE bottles  Sterilized | 120 ml | Thiosulfate  40C in dark | 6 hrs |

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

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

**B2.1 In situ Water Quality Monitoring**

1. **Equipment/Instrument Calibration**

Prior to field use, the multi-parameter or individual meters will be calibrated in accordance with the manufacturer’s instruction manual. If not 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 (DI) and tap water in squirt bottles
* Calibration cups and standards
* Kim wipes or paper towels
* Holosteric barometer or elevation chart to use for calibration
* NIST-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 (1000 μS/cm) conductivity standard
* Sodium sulfite solution (0% dissolved oxygen)

The following items will also be needed for recording measurements

* Field Measurement form
* Pencils (for data forms)

**Method: Equipment calibration (for multi-parameter unit or individual units)**

Temperature

The accuracy of the sensor will be checked against a thermometer that is traceable to the National Institute of Standards and Technology (NIST) at least once per sampling season

pH

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

Calibrate the pH meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer’s instructions and existing Standard Operating Procedures (SOP). Ideally, a quality control solution (QCS) should be used that is similar in ionic strength to the water samples you will be measuring.

*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 Kim-wipes or paper towel. 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 lab sheet.

Dissolved Oxygen

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

Calibrate the DO 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 DO 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 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 lab sheet.

Conductivity

*Calibration standard required*: 1 mS/cm (1000 μS/cm) conductivity standard

Calibrate the conductivity meter prior to each sampling event. Calibrate the meter in accordance with the manufacturer’s instructions. Ideally, a QCS solution should be used that incorporates the entire expected conductivity range

*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 Kim-wipes or paper towel. 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 lab sheet.

Using the Data Logger

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

1. **Multi-parameter unit deployment and grab sample collection**

A hydrographic profile at each site will be obtained at water depth greater than or equal to 2 meters. 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 then 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. For stations where multi-parameter unit or single-parameter units are used: Estimate the total water depth at the sampling site by lowering the depth sounding line (marked in feet) to the bottom of the river and counting the number of taped one-foot marks on the cable. If possible, sensor measurements will be collected during the downcast from near surface (approximately 0.5-1.5 mm) to near bottom (about 0.5 m off the bottom) or along a hydrographic profile.
2. 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 no water in the stream, record as “No Flow”.

[TIP]

As a rule to thumb do not wade in to sample if water depth (ft.) x velocity (fps) > 10. Readings can be taken at the bottle sampling site if the river is shallow or from a bridge near the bottle 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 ensure that a sample taken from the bank or from a bridge is in the flow and representative of the larger area. If a representative location cannot be found, do not take sample.

**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 meter, take the measurements at mid-depth. Avoid the probe contacting bottom sediments, as dissolved oxygen readings in the sediments are very low. If conductivity readings are zero, check that the probes are covered with water.
4. 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 “In-situ Readings” section of the River Observation field sheet. Record the data on the data logger (as described below). Record the measurements on the Field Measurement Form.

**B2.2 Nutrients**

**[THIS WILL APPLY WHEN USERS SELECT TOTAL N, AMMONIUM-N, NITRATE-NITRITE-N, TOTAL P, AND ORTHOPHOSPHATES]**

Supply list for collecting samples

* Nitrile gloves
* 4 L sample container
* 2 L amber Nalgene bottle
* Cooler with ice
* Dry Ice
* Plastic electrical tape
* DI water

For recording measurements

* Sample Collection Form
* Sample label with pre-printed Sample ID
* Clear tape strips
* Sample Collection Form
* Pencils (for data forms) Fine tipped indelible markers

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

**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 in refrigerator at 40C 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).
4. If samples cannot be delivered to the laboratory within 6-8 hours following collection, acid preservation may be required. S**ee Method: Processing Nutrient Samples**

**Method: Processing Nutrient Samples**

1. To acidify samples, put disposable gloves and safety glasses on.
2. Remove acid bottle from ziploc bag, and carefully draw approx. 1 ml. of 9N H2SO4 per 250 mls. (e.g., 2 mls. for 500 ml sample) and dispense into sample to achieve sample pH < 2.
3. Cap sample and mix thoroughly. Carefully recap acid and discard used pipette into separate acid-refuse bag. Place samples back on ice.

**B2.3 Chlorophyll-a**

Supply list for collecting samples

* Nitrile gloves
* 250 L amber Nalgene bottle
* Cooler with ice
* Dry Ice
* Plastic electrical tape
* DI water

For recording measurements

* Sample Collection Form
* Sample label with pre-printed Sample ID
* Clear tape strips
* Sample Collection Form
* Pencils (for data forms) Fine tipped indelible markers

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

**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, samples will be stored in refrigerator at 40C and processed 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 sheet.
3. Pour the measured sample into the clean filter funnel and filter with a vacuum pump (electric pump or by hand until the vacuum is 15" of vacuum units). Filter a minimum of 500-ml of 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 milliliter.
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.
7. When all samples have been filtered, the drying box is plugged in. 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 and prepare for delivery to the laboratory or freeze.

**2.4 Chlorides**

**[THIS WILL APPLY WHEN USER SELECTS CHLORIDES]**

Supply list for collecting samples

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry Ice
* Plastic electrical tape
* DI water

For recording measurements

* Sample Collection Form
* Sample label with pre-printed Sample ID
* Clear tape strips
* Sample Collection Form
* Pencils (for data forms) Fine tipped indelible markers

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

**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 in refrigerator at 40C and process within 28 days.

**2.5 Turbidity**

**[THIS WILL APPLY WHEN USER SELECTS TURBIDITY]**

Supply list for collecting samples

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry Ice
* Plastic electrical tape
* DI water

For recording measurements

* Sample Collection Form
* Sample label with pre-printed Sample ID
* Clear tape strips
* Sample Collection Form
* Pencils (for data forms) Fine tipped indelible markers

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

**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 in refrigerator at 40C and process within 48 hours.

**2.6 Total Suspended Solids**

**[THIS WILL APPLY WHEN USER SELECTS TOTAL SUSPENDED SOLIDS]**

Supply list for collecting samples

* Nitrile gloves
* 4 L sample container
* Cooler with ice
* Dry Ice
* Plastic electrical tape
* DI water

For recording measurements

* Sample Collection Form
* Sample label with pre-printed Sample ID
* Clear tape strips
* Sample Collection Form
* Pencils (for data forms) Fine tipped indelible markers

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

**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 in refrigerator at 40C and process within 7 days.

**2.7 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 bottle. A screening test is conducted in the field using dipsticks. If 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
* Sample Collection Form
* pencils (for data forms) and Fine tipped indelible markers for labels

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure [SECTION WILL APPEAR]**

**Field Method: Collecting duplicates and field blanks [SECTION WILL APPEAR]**

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

1. Pour 1-2 ml of the sample from the HDPE 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 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.
9. 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.

**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 six hours of collection.

**Pre-sampling site assessment [SECTION WILL APPEAR]**

**Field Method: General Bottle Sampling Procedure**

**For wadeable rivers and streams using grab samples**

1. Wade in carefully, moving from downstream to upstream until you get to the main the 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.
3. Put on sterile, nitrile gloves.
4. Lower the uncapped, inverted 120 ml sample bottle to a depth of 6 inches below the water surface, avoiding surface scum, vegetation, and substrates.
5. Point the mouth of the container away from the body or boat. Right the bottle and raise it through the water column, allowing bottle to fill completely.
6. 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.
7. 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).
8. Add the sodium thiosulfate tablet, cap, and shake bottle 25 times

**Non-wadeable Rivers and Streams**

If it is not safe 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 the sample collection rod 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 40C 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.

**Elevated drop/bridge grabs (Rivers and Streams >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, 1000 ml NM, 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 inside of bottles. Lower slowly to water surface and gently plunge into water to approx. 6 inches below water surface and allow bottles to fill.
6. Observe 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 1000 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 40C immediately.

TIPS OR NOTES?

* Resample if the sampler over-fills the container or contact had been made to sample. Always take an extra sterile bacteria sample bottle for use in cases where a bacteria sample bottle has been wasted, and re-sampling is needed. It is important to document the samplers’ expert opinion on the chain of custody when a site is suspected of having a high bacteria concentration. Informing the bacteria analyst when the site is suspect provides information on how the sample should be treated during analysis.

**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 six 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 waterbody. Record these disturbances on the Site Assessment Form. If samples are not delivered within 6 hours to the laboratory, 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 PP tube
* Filtration apparatus with collection flask
* Sterile filter holder, Nalgene 145/147
* Vacuum pump (electric pump may be used if available)
* Sterile phosphate buffered saline (PBS)
* Osmotics 47 mm polycarbonate sterile filters
* Sterile disposable forceps
* Petri dishes (60 x 15, disposable)
* 2 sterile microcentrifuge tubes containing sterile glass beads
* 1 additional sterile microcentrifuge tube if collecting filter blank
* Bubble bag
* Zip-top bag
* Dry ice
* Cooler

For recording measurements

* Sample Collection Form
* Soft (#2) lead pencils for recording data on field forms
* Fine-tipped indelible markers for filling out sample labels
* Fecal Indicator sample labels (2 vial labels and 1 bag label)
* Filter blank label if collecting filter blank

The sample must be filtered and frozen within six 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 (PBS), 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 filter box to base of opened Petri dish. Close filter box and set aside.
5. Remove the pre-loaded cellulose nitrate (CN) filter from funnel and discard. Be sure to leave the support pad in the filter funnel.
6. Load filtration funnel with 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 inches of Hg 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 5 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.

**[Use for pre-sampling site assessment for nutrients, chlorophyll, turbidity, TSS, chlorides, bacteria and microcystins]**

**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 to thumb (USGS) do not wade in to sample if water depth (ft.) x velocity (fps) > 10. Where wading into the flow is not possible for the chosen site and no other 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.

**[Use General Bottle Sampling Procedure (Wadeable Rivers and Streams) for nutrients, chlorophyll, turbidity, TSS, chlorides, bacteria and microcystins]**

**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 the 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 (the water moving towards 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 proceed to 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 the hand. Open sampling container about 6 inches 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 40C immediately.

**[Use General Bottle Sampling Procedure (Non-wadeable Rivers and Streams) for nutrients, chlorophyll, turbidity, TSS, chlorides, bacteria and microcystins]**

**Non-wadeable Rivers and Streams**

If it is not safe 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 the sample collection rod 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 and 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 40C 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.

**[Use for General Bottle Sampling Procedure (Elevated drop/bridge) for nutrients, chlorophyll, turbidity, TSS, chlorides, bacteria and microcystins]**

**Elevated drop/bridge grabs (Rivers and Streams >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. 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 new plastic baggie. Do not rinse the inside of the bottles.
5. Lower slowly to water surface and gently plunge into water to approx. 6 inches below water surface and allow bottles to fill. Observe 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 40C immediately.

**[Use for Colleting Duplicates and Field Blanks for nutrients, chlorophyll, turbidity, TSS, chlorides, bacteria and microcystins]**

**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.

NOTES OR TIPS?

* If sediments are disturbed by the sampler at any time, wait until the disturbance has abated before taking any samples (Sampler’s position 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).
* If water depth is less than 18 inches approx. water depth, do not take samples using weighted basket sampler. Find a spot for taking wade-in grab samples.

**B.3 SAMPLE HANDLING AND CUSTODY**

+++ IF 4 < 3 +++

Identify the process of sample handling, custody in the field, laboratory and transport, taking into account the nature of the samples, the maximum allowable holding time before extraction or analysis and available shipping options and schedules. The process should include:

* Sample Identification Procedures
* Chain-of-Custody Procedures
* Field Sample Custody/Tracking
* Laboratory Sample Custody/Tracking

+++ END-IF +++

**Sample identification system**

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:**

* Ensure all samples are appropriately preserved and packaged for transport.
* If obtainable samples are missing, the [crew leader] 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).
* Ensure that all samples are labeled.
* Make sure all labels are complete, legible, accurate, and consistent.
* Review the labels and forms to ensure consistent sample ID information was utilized.
* Inspect the integrity of each sample container; be sure there are no leaks. Make sure that all sample containers are properly sealed.

**Chain-of-Custody Procedures**

+++ IF 4 < 3 +++

Instructions: Use a Chain-of-Custody form and describe the procedure used to ensure every person who handles transports or accepts samples sings the form with date and time.

+++ END-IF +++

The sampling crew team leader 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. Crews include copies of all tracking forms in the coolers when they send samples to the labs.

[SPACE FOR FREE TEXT]

**Field sample custody/tracking procedures**

+++ IF 4 < 3 +++

Describe the custody and tracking procedures that will be used from sample collection to packaging to shipment to delivery to the laboratory. Describe the type of shipment or carrier used. State where samples will be kept and preserved while transporting to the laboratory.

+++ END-IF +++

[SPACE FOR FREE TEXT]

**Laboratory sample custody/tracking procedures**

+++ IF 4 < 3 +++

Describe the custody and tracking procedures that will be used from sample to preparation to analysis to archiving to disposal. You may provide the SOP used by the laboratory.

+++ END-IF +++

[SPACE FOR FREE TEXT]

**B4 ANALYTICAL METHODS**

+++ IF 4 < 3 +++

The table lists the standard methods approved for application to samples under this QAPP. If any additional or alternative methods are used, include in the table.

+++ END-IF +++

**Table 4.1 Approved Analytical Methods**

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Parameter** | **Method #1** | **Method #2** | **MDL (mg/l unless stated)** | **Lab Name** |
| Turbidity | EPA 180.1 | SM 2130-B | 0.2 NTU |  |
| Chlorophyll a | EPA 445.0  modified | SM 10200-H | 1 μg/l |  |
| Total Nitrogen (TN) | EPA 353.2 (modified) | SM 4500-N USGS I-4650-03 | 0.05  0.05 |  |
| Nitrate-Nitrite-N | EPA 353.1 | SM 4500-NO3 | 0.02 |  |
| Ammonia-N | EPA 350.1 | SM 4500-NH3 | 0.02 |  |
| Total Phosphorus (TP) | EPA 365.2 | SM 4500-P  USGS-1-4650-03 | 0.01 |  |
| Orthophosphate | EPA 365.5 | SM 4500-P  USGS-1-4650-03 | 0.01 |  |
| Chloride |  | SM 4500-CL |  |  |
| Total suspended solids | EPA 160.2 | SM 2540-D | 1.0 |  |
| Microcystins | Abraxis Strip Test (field) | EPA 544  EPA 546 | 0.5 ug/l |  |
| E. coli | EPA 1603 | SM 9223B | 5 cfu/100 ml |  |

**B.5 FIELD AND ANALYTICAL LABORATORY QUALITY CONTROL**

+++ IF 4 < 3 +++

Instructions: Identify the number and types of field and lab QC samples. Complete a separate table for each analytical group as needed. Include the field QC samples that will be collected and sent to the laboratory.

+++ END-IF +++

Specific quality control measures are listed in Table 5.1for field measurements and observations. Project sampling will include appropriate field and laboratory quality control samples to assess general data quality issues, as well as specific data quality objectives.

Field quality control samples will be taken for 10% of all water quality samples taken per sampling trip. Example numbers of QC samples required to meet an approximately 10% rates are as follows:

* 1-10 samples taken, 1 QC sample is processed.
* 11-20 samples taken, 1-2 QC samples are processed.
* 21-30 samples taken, 2-3 QC samples are processed.

**Field Duplicates**

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

**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 deionized 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 5.1 Quality Control Measures

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Sample type** | **Instrument/**  **Parameter** | **Accuracy Checks** | **Precision Checks** | **% Field QC**  **Samples** |
| Multi-parameter instrument | All | Pre- and post-survey calibration including “zero” DO std. check | 3-5 minute of stable readings logged or recorded | Verify repeatability in the field |
| Single probe instruments | Thermometer | Compare with certified thermometer | Field duplicates | 10% |
|  | Conductivity | Field blanks, QC std | Field duplicates | 10% |
|  | DO | Compare w/ Winkler titration method results | Field duplicates | 10% |
|  | pH | Blind audit samples | Field duplicates | 10% |
| Grab water samples | TN, NH3-N, NO3-NO2-N, TP, ortho-P | Field: blanks  Lab: analysis of lab-fortified matrix (spiked samples) and/or lab QC std. | Field: duplicates  Lab: duplicates | 10% |
|  | Chlorides | Field: blanks  Lab: analysis of lab-fortified | Field: duplicates  Lab: duplicates | 10% |
|  | Turbidity | External audit/QC std, distilled water  Lab: blanks | Field: duplicates  Lab: duplicates | 10% |
|  | Total Suspended Solids | External audit/QC std, distilled water  Lab: blanks | Field: duplicates  Lab: duplicates | 10% |
|  | Chlorophyll-a | Commercial audit samples | Field duplicates | 10% |
|  | Microcystins |  | Field duplicates | 10% |
|  | E. coli | Colilert audit samples | Field duplicates | 10% |

**Quality Control Procedures: Field Operations**

+++ IF 4 < 3 +++

Describe the steps taken by the [field crew] as part of the [monitoring program] QC procedures.

+++ END-IF +++

[SPACE FOR FREE TEXT]

Table 5.2 Field quality control: in situ parameters.

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **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. | |

Table 5.3 Field 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. 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. |

Table 5.4 Sample Field Processing Quality Control: Chlorophyll–a

[TEMPLATE TO BE PROVIDED BY APP] [Table rows to be included per parameters selected by user]

|  |  |  |
| --- | --- | --- |
| **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 |

Table 5.5 Data Validation QC for water chemistry

|  |  |
| --- | --- |
| **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 |

**Table 5.6 Sample Collection and Field Processing Quality Control: Fecal Indicator**

[TEMPLATE TO BE PROVIDED BY APP] [Table rows to be included per parameters selected by user]

|  |  |  |
| --- | --- | --- |
| **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 5.7 Data Validation Quality Control: E. coli.

[TEMPLATE TO BE PROVIDED BY APP] [Table rows to be included per parameters selected by user]

|  |  |  |  |
| --- | --- | --- | --- |
| **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 |
| **Data processing & review** | | | |
| 100% verification and review of qPCR data | All qPCR amplification traces, raw and processed data sheets | All final data will be checked against raw data, exported data, and calculated data printouts before uploading to WQX. | Second tier review by contractor. |

Table 5.8 Sample Field Processing Quality Control: Microcystins

[TEMPLATE TO BE PROVIDED BY APP] [Table rows to be included per parameters selected by user]

|  |  |  |
| --- | --- | --- |
| **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 5.9 Data validation Quality Control for 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**

+++ IF 4 < 3 +++

Describes the steps taken to ensure instrument/equipment are inspected and tested periodically. Provide any maintenance and calibration requirements for the equipment used for the [monitoring program]. State how the calibration information will be documented.

+++ END-IF +++

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. Examples include: multi-parameter probes ‐ annual (or as needed) maintenance and calibration check by manufacturer or certified service center. 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 SOPs for instrument maintenance and calibration.

Table 6.1 Typical Instrument/Equipment Inspection and Testing Procedures

[Table rows to be included per parameters selected by user]

|  |  |  |  |  |
| --- | --- | --- | --- | --- |
| **Equipment** | **Inspection frequency** | **Type inspection** | **Maintenance, Corrective Action** | **Person Responsible** |
| Sample bottles | Before each use | Visual for integrity, cleanliness | Acid washed prior to use (or clean-certified from manufacturer or lab) |  |
| Filtering apparatus | Before each sue | Proper functioning, clean storage | Spare filters, syringe |  |
| Meters | Before each use | Battery life, DO membrane | Spare batteries, spare membranes |  |
| GPS | Before each use | Battery life | Repair, replace, spare batteries on hand. |  |
| Sampling device | Before each use | Visual for integrity, cleanliness | Repair, replace if necessary |  |

**B7 FIELD EQUIPMENT/MAINTENANCE, INSPECTION AND CALIBRATION**

+++ IF 4 < 3 +++

Describes how continued quality performance of equipment and instruments will be ensured prior to the field season and each field trip as needed.

end of if

+++ END-IF +++

[SPACE FOR FREE TEXT]

**Pre-measurement Instrument Checks and Calibration**

+++ IF 4 < 3 +++

Describe what pre-measurement checks and calibrations are performed prior to the field season and each field trip as needed.

+++ END-IF +++

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 global positioning system (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 hand-held 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 meter (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 meter**

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

Table 7.1 Instrument Calibration Procedures

[Table rows to be included per parameters selected by user]

|  |  |  |  |
| --- | --- | --- | --- |
| **Instrument** | **Inspection and Calibration Frequency** | **Standard of Calibration Used** | **Corrective Action** |
| Calibrated line | Annually | Tape measure | Recalibrate or replace |
| Multi-parameter probe meter | Before each sampling run | Std. solutions DO probe compare w/Winkler titration | According to manufacturer’s instructions. DO: replace membrane or correct probe |
| Thermometer | Annually | NIST certified thermometer | Replace or provide correction factor |
| Conductivity meter | Before each sampling run | Use certified inspection stds | Adjust and recalibrate |
| Turbidity meter | Before each sampling run | Known standards | Adjust according to manufacturer’s recommendations |
| pH meter | Before each sampling run | pH buffers 4.01 and 7.00 or external stds (4,7,10) | Adjust instrument, clean electrodes, replace if needed |

External standards refer to standards of reliable quality obtained from reputable commercial or other supplier, Known standards refer to those where the value is known before calibration.

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**Post-Measurement Calibration Check**

+++ IF 4 < 3 +++

Describe what post-measurement checks and calibrations are performed prior to the field season and each field trip as needed.

+++ END-IF +++

**GUIDANCE**

**B7.2.1 Multi-Parameter Sonde**

After all *in situ* measurements have been completed for the sampling day, a post-measurement calibration check of the multi-parameter sonde 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 meter may need service and data collected since the last successful calibration and post-measurement calibration check should be flagged. Discontinue use of any meter that is not functioning properly.

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

+++ IF 4 < 3 +++

Describe the process for consistently checking, testing and maintaining instruments and equipment for proper functioning.

+++ END-IF +++

**GUIDANCE**

Maintenance shall occur as needed. Records of equipment inspection, maintenance, repair and replacement shall be kept in a logbook. In addition to following a manufacturer’s recommendations, group-specific SOPs for instrument maintenance and calibration shall be developed and followed.

**B9 DATA ACQUISITION REQUIREMENTS**

+++ IF 4 < 3 +++

Describe any existing data to be obtained from external sources as well as plane to evaluate any limitations on its intended use for the current project.

+++ END-IF +++

GUIDANCE

Such data may include:

* + Existing sampling and analysis data form a previous project
  + Photos or maps
  + Published literature
  + Information from public databases
  + Weather or GPS location date.
  + USGS streamflow readings
  + Associated source information must be identified.

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 sources,

* Title of document or description name of the information
* Source of information
* Notes on quality of data, including whether it has a QAPP or some other means of demonstrating quality of data
* As applicable, a statement on planned restrictions in use of the data due to questions about data quality.

Table9.1 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**

+++ IF 4 < 3 +++

Provide an overview of the management of data generated throughout the project.

+++ END-IF +++

GUIDANCE

Field samples shall record data on field sheets, review them, sign, and submit to the Project Field Coordinator. The Project Field Coordinator will review the sheets 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 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 sheers for accuracy. The original data sheets will be stored in the organization’s office. Electronic backups and copies of data sheets 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.

Table 10.1 Data management, review, validation, verification process [TEMPLATE TO BE PROVIDED]

|  |  |  |
| --- | --- | --- |
| **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 |
|  |  |  |
|  |  |  |

**B. FRESH WATER / BENTHIC DATA GENERATION AND ACQUISITION**

**B1. Sampling Design**

AquaQAPP should recommend the following freshwater WQ parameters be included with benthic monitoring: temperature, pH, DO, TSS or turbidity, and conductivity. Chl*a*  and microcystin are optional.

TIP: Ideally, site selection criteria would be 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).

**B1 Sample site selection** **– AquaQAPP concern = source impact**

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 (>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).

**B1.2 Sample site (reach) selection and assessment** **– AquaQAPP concern = general benthic health**

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 NPS pollution or other pertinent information) during the time of reconnaissance will be noted and recorded in a field notebook.

**B2. Sampling Methods** **(All FW WQ AquaQAPPs)**

**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 logged with a brief description.

**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 feet)
* Orange
* Net
* Stopwatch
* Calculator

1. Measure off 25 feet along the bank of a straight section of stream. The stream stretch chosen for the measurement of discharge should be straight (no bends), at least 6 inches deep, and should not contain an area of slow water such as a pool. Unobstructed riffles or runs are ideal. Stretch string across each end of the 25- foot 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 feet instead of 25). Repeat the measurement at least two more times, more for greater accuracy. Average the results (sec).
3. Calculate flow velocity as distance traveled (ft) divided by the average amount of time (sec) it took the float to travel that distance.

**B2. Sampling method – viewing bucket**

**(This section for viewing bucket and parameters: benthic algal biomass)**

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 and described in EPA’s Rapid Assessment Protocol) 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.

**Field Equipment for Benthic Algal Biomass Assessment**

Meter stick

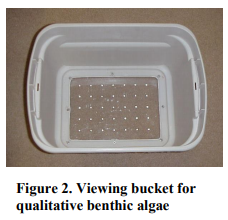
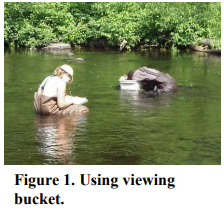
Pencil

Field Sheet

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 water tight seal so that no water enters the bucket when it is partially submerged. Mark 50 dots in a 7 x 7 grid on the top surface of the acrylic sheet with a waterproof black marker. Add another dot outside the 7 x 7 grid to make the 50 dot grid.]

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 2).
3. At each location, the recorder notes the transect and sample number, and instantaneous DO, temperature, pH, and flow velocity using field meters.
4. The viewer then immerses the viewing bucket in the water (Figure 3). 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 2.** Using viewing bucket **Figure 3.** 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, waterbody name, and date written in large, thick letters, followed by a photograph of the viewing area.
2. To characterize macroalgal biomass:
   1. Observe the bottom of the stream through the bottom of the viewing bucket and count the number of dots that occur 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 sheet.
   2. Measure and record the maximum length of the macroalgae.
   3. If two or more types of macroalgae are present, count the dots, measure, and record information for each type of macroalgae separately.
   4. 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.
3. To characterize extent of suitable substrate:
   1. Record the number of dots under which there is gravel greater than 2 cm in diameter.
4. To characterize microalgal cover:
   1. 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 sheet. The recorder will make a hash mark under the corresponding category.
   2. 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 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 mm to 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:
   1. Convert the number of hash marks for each category to a numeral, and record that number under the category. (See sample data sheet [ME DEP Viewing Bucket Survey Data Sheet](https://www.maine.gov/dep/water/monitoring/biomonitoring/materials/fieldsheet_viewingbucket.pdf))
   2. 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, name of sampler. The algal sample will be held on ice for transport to [observation location] within 24h.

**B4. Analytical Methods – algal biomass**

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

Density of algae on substrate may be determined using the following statistics [(EPA RBP AR1164)](https://www3.epa.gov/region1/npdes/merrimackstation/pdfs/ar/AR-1164.pdf):

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 = ??

**B2. Sampling Methods – Kick Sampling**

**(This section for kick samplingt and parameters: macroinvertebrates)**

**Info/tips:**

SAMPLING NET TYPES (all use a standard screen mesh size of 500 μm)

* Kick net: Dimensions of net are 1 meter (m) x 1 m attached to 2 poles and functions similarly to a fish kick seine. Is most efficient for sampling cobble substrate (i.e., riffles and runs) where velocity of water will transport dislodged organisms into net. Designed to sample 1 mof substrate at a time and can be used in any depth from a few centimeters to just below 1m (Note -- Depths of 1m or greater will be difficult to sample with any gear).
* D-frame dip net: Dimensions of frame are 0.3 m width and 0.3 m height and shaped as a “D” where frame attaches to long pole. Net is cone or bag-shaped for capture of organisms. Can be used in a variety of habitat types and used as a kick net, or for “jabbing”, “dipping”, or “sweeping”.
* Rectangular dip net: Dimensions of frame are 0.5 m width and 0.3 m height and attached to a long pole. Net is cone or bag-shaped. Sampling is conducted similarly to the D-frame.

**Method Summary**

Benthic macroinvertebrate samples will be collected using a [type] net with 500 µm mesh openings. Composite samples from stations at 11 transects multiple sites are preserved in the field with 95% ethanol, then sorted and sent to the laboratory for identification.

**Equipment and Supplies**

* [type] net, 500 μm mesh
* Timer or stopwatch
* Soft nylon brush
* Forceps
* Small spatula, spoon, or scoop to transfer sample
* Sample jars (1L HCPE plastic suitable for use with 95% ethanol)
* 95% ethanol, properly stored and labeled
* Wash bottle (1L, labeled “STREAM WATER”)
* Cooler (with absorbent material for transporting ethanol and samples)
* Plastic electric tape
* Scissors
* Blank and completed labels
* Indelible-ink markers
* #2 pencils
* Clear tape
* Sample collection field sheets
* Blank labels on waterproof paper for internal sample labels

**Sampling Procedure**

Stream sampling site will be a wadeable, 100m-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. Any large debris items (rocks, sticks, etc.) caught in the net are rinsed in the net and returned 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 having 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 approximately two square meters.
8. In streams where the riffles within the reach are inadequate to allow for a two-square-meter composite, other productive habitats may be sampled by jabs into snags, or rubbing substrates. In such cases, notes will be recorded on the field sheets indicating the number of kicks or jabs in each habitat category that contributed to the composite sample.
9. Any debris (gravel, rocks, sticks, etc.) caught in the net will be rinsed in the net and returned to the stream, once any macroinvertebrates clinging to it have been removed. As much gravel as possible will be cleaned and removed so that organisms do not get damaged. The residue in the net will be placed in a container with enough denatured 95% ethanol added to cover the residue.
10. A label (sample provided) filled in with a #2 pencil will be placed inside the sample container, and a duplicate label with the words “preservative: denatured 95% ethanol,” will be affixed to the outside of the container as well.

**B2. Sampling Method – Rock Baskets**

**(This section for rock baskets and parameters: macroinvertebrates)**

### 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 6 to 8 weeks. Composite samples from multiple sites are preserved, then sorted and sent to the laboratory for identification.

**Equipment list**

* Rock-filled wire or mesh baskets, three per sampling site
  + Substrate: clean, washed cobble (1 to 3in diameter) or #2 roofing stone
  + Basket mesh size: 1.5 to 2.5cm
  + Fill weight: 7.25 + 0.5kg
* Sieve bucket with 500 μm mesh
* Soft nylon brush
* Forceps
* Small spatula, spoon, or scoop to transfer sample
* Sample jars (1L HCPE plastic suitable for use with 95% ethanol)
* 95% ethanol properly stored and labeled
* Wash bottle (1L, labeled “STREAM WATER”)
* Cooler (with absorbent material for transporting ethanol and samples)
* Plastic electric tape
* Scissors
* Blank and completed labels
* Indelible-ink markers
* #2 pencils
* Clear tape
* Sample collection field sheets
* Blank labels on waterproof paper for internal sample labels

**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. The location should be approached 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 days +/- four days exposure period may be used.
4. At the completion of the exposure interval, approach from downstream to retrieve the basket(s). Each basket is retrieved by pressing a kick-net tightly against the streambed along the basket’s downstream edge. The basket is then moved carefully into the net before lifting it through the water column. Where the water is deep enough to make this procedure difficult or impossible, a basket made with 500 μm netting on its bottom surface will be used to prevent the loss of organisms through the bottom of the sampler as it is raised to the surface.
5. Vegetation or debris snagged on the outside of the recovered sampler is removed, taking care to avoid jarring the sampler. and the basket is placed in a large sieve bucket or tub of water. Empty net contents into the bucket as well. The sampler is then opened, emptied into the bucket, rinsed inside the bucket until free of any organisms or adhering material, and set aside. Each rock is similarly rinsed and set aside. The material remaining in the bucket is then sieved.
6. Wash the contents of the sieve to one side by gently agitating the sieve in the water. Transfer the residue to a labeled 1L sample bottle (see Step 15) 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. A label (sample provided) filled in with #2 pencil will be placed inside the sample container, and a duplicate label with the words “preservative: denatured 95% ethanol,” will be affixed to the outside of the container as well.

**B2 Sample processing (sorting) – Kick sampling & rock basket**

**(This section for kick sampling and rock basket and parameters: macroinvertebrates)**

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, and 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; and,
* the aquatic life stages of Insecta except Hemiptera and adult Coleoptera other than Elmidae.

Those macroinvertebrates excluded from the above list are not used for one of three reasons: either 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 (ref.).

**Equipment list**

 70% ethanol[[1]](#footnote-1)

 Nitrile or latex gloves

 500-micron mesh sieve

 2 white plastic trays, one marked with a 3in by 3in grid

 250ml bottle (preferably with no lip)

 Sufficient light source

 Waste bucket

 Forceps

 Label tape

 Resistall label paper

 Pencils or India ink pens

 Magnifying glass

**How to pick your sample**

1. Prepare a clean jar with a label. The label needs an identification code that will help us to know where the sample came from and also record the sample in our computer records. The code is a single line of numbers and letters consisting of the following information:
   1. Site number
   2. Field collection date
   3. Field sampler initials
   4. Picking date – the date on which you are picking the sample
   5. Picker initials – three letter set of initials, like the field sampler initials, for the person who is picking the sample
2. Fill the jar with 70% ethanol.
3. Empty the contents of the field-sampled jar into 500-micron mesh sieve then thoroughly rinse the sample to remove ethanol and fine debris.
4. Collect and dilute 90% ethanol from field sample in a 4:1 ratio with tap water before disposal
5. You should pick out any large debris and look it over carefully to check for macroinvertebrates before discarding it.
6. You can then empty the remaining contents of the sieve into the first (non-marked) tray.
7. 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.
   1. Looking through one square of the grid at a time you should pick out macroinvertebrates with your forceps and put them into the labeled jar. Do not use your bare hands!
   2. Once you have searched every square of the grid you should shake your tray gently to mix up the remaining contents.
   3. Again, search every square of the grid to make sure you have found all the macroinvertebrates.
   4. Any water and debris remaining on the tray can then be discarded.
   5. Move the next portion of the sample onto the gridded tray and search it.
8. Once you have finished picking through the entire sample, make sure the contents of your labeled jar are completely covered in 70% ethanol.
9. Copy out the identification code from the label of your jar onto waterproof paper using a pencil.
10. Then put this new label inside the jar and close the jar securely.
11. The original sample jar can be rinsed and reused for future sampling.
12. Store all full jars at room temperature in a secure location.

**B3. Sample Handling and Custody Requirements – kick sampling & rock basket methods**

**AquaQAPP parameter: macroinvertebrates**

Macrointertebrate 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 drop off. 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 2-day express delivery if necessary for delivery to 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– kick sampling & rock basket methods**

**AquaQAPP parameter: macroinvertebrates**

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 4/26/19). 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 Requirements– kick sampling & rock basket methods**

**AquaQAPP parameter: macroinvertebrates**

Table 1 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 ten transects sampled for each reach. Duplicate samples are preserved in a separate sample bottle marked “duplicate” and with all other information regarding station location remaining the same. Duplicate samples will be used in the calculation of precision of the benthos data.

Macroinvertebrate Sampling

|  |  |  |  |
| --- | --- | --- | --- |
| **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.

**Quality Control for sorting/picking**

Ten percent of the sorted samples in each lot should be examined by laboratory QC personnel or a qualified co-worker. (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 less 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, a second 10% of the sample lot will be checked by the QC worker. 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.

**B2. Sampling Method – Physical Habitat Assessment**

**Physical Habitat Assessment and parameter: Stream characteristics**

AquaQAPP Information: The procedure for assessing physical habitat quality included here is an integral component of the final evaluation of impairment. The matrix used to assess habitat quality is based on key physical characteristics of the waterbody and surrounding land, particularly the catchment of the site under investigation. Physical characterization includes documentation of general land use, description of the stream origin and type, summary of the riparian vegetation features, and measurements of instream parameters such as width, depth, flow, and substrate. All of the habitat parameters evaluated are related to overall aquatic life use and are a potential source of limitation to the aquatic biota.

**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 Barbour et al., 1999. (EPA Rapid Bioassessment Protocol, Section 5.2)

**Equipment list**

* Field meter (for water chemistry and flow velocity)
* Meter stick, meter tape, or range finder
* Digital camera
* [choose from/adapt these:]
  + [Data sheet](https://www.maine.gov/dep/water/monitoring/biomonitoring/materials/fieldsheet_algae_EPA_physical_characterization.pdf)  (EPA)
  + [Field data sheets](https://www.des.nh.gov/organization/divisions/water/wmb/biomonitoring/documents/data_sheets.pdf) (NHDES)
* Clipboard
* Pencils or waterproof pens

Upstream/downstream “arrows” or signs forphotographing and documenting sampling reaches

* GPS unit

**Method description**

1. The habitat assessment is performed 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 each field data sheet and habitat assessment form. It is best for the investigators to obtain a close look 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 and Water Quality Field Data Sheet**. Sketch a map of the sampling reach on the back of this form.
4. Complete the **Habitat Assessment Field Data Sheet**, in a team of 2 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 2 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. The categories are (descriptions from MEDEP):

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.
2. a) **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.
3. b) **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 than a stream that has a variety of substrate types.
4. a) **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.
5. b) **Pool Variability (high-gradient streams):**  This rates the overall mixture of pool types found in streams, according to size and depth. There are 4 basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream will support a wide variety if 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.
6. **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.
7. **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.
8. **Channel Alteration:** Channel alteration is an assessment of the degree of diversion from the natural course of the water body by man-made 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.
9. a) **Riffle Frequency (low-gradient streams):**  Riffle habitat is considered to be the in-stream 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.
10. b) **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.
11. **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 in-stream water temperatures, and cause community shifts from pollutant sensitive aquatic species to pollutant tolerant ones. Poor streamside bank conditions usually coincide with poor in-stream habitat.
12. **Bank Vegetative Protection:** Stream side vegetation is one of the principal factors which protects the streambank from erosional processes, provides shade and protective cover for aquatic life, and provides a significant food source to in-stream 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.
13. **Riparian Vegetative Zone Width:** This habitat quality parameter assesses the width of naturally occurring vegetation between the water body and the area of man-made 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 eighteen meters is considered optimal; additional widths will in most cases not result in additional protection or attenuation of pollutants.

##### B3, B4 not applicable to this method

##### B5. Quality Control – Physical Habitat Assessment

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

The following B sections are relevant to all methods & parameters

**B6 – Instrument/Equipment Testing, Inspection, and Maintenance Requirements**

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

**Materials**

* 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:

(1) Always wear gloves and safety goggles when using disinfectant, and avoid contact with exposed skin, clothing, vehicle upholstery and/or other fabric.

(2) New bleach solution must be made up daily. New quaternary ammonia solution should be made up every 2-3 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.

(3) 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).

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 waterbody or along the shore. All plant fragments must be removed before equipment is transported to another waterbody.
2. 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 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 feet 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 feet from open water and remove mud and other debris, by washing with soap and water and rinsing as described in Level 2 cleaning 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 3 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 waterbody 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.
   1. Air dried for several months, in direct sunlight for part of the time, if possible, or
   2. 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 – all parameters/methods**

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 sheets and chain-of- custody forms.
* coordinating the transfer of all samples from the field to laboratories for analysis.

**Table 1. Supplies, Acceptance Criteria, and Responsibility for Critical Field Supplies** ***– all parameters/methods***

| **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. | Chief Scientist |
| 95% ethanol | Visually inspected for proper labeling, expiration dates, appropriate grade. | Chief Scientist  Laboratory Staff |
| Sampling equipment | Visually inspected for obvious defects, damage, and contamination. | Chief Scientist |
| Navigation instruments, digital camera | Functional checks to ensure proper calibration and operating capacity. | Vessel Captain  Chief Scientist |

**B9 – Data Acquisition Requirements** (Non-direct Measurements**) *– all parameters/methods***

Non-direct 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

* Mass GIS program or CZM’s MORIS maps
* Prior reports specific to the area.
* Results of other studies including water quality monitoring and benthic survey data.
* Peer-reviewed publications of surveys completed in the area or system of interest.

B10. Data Management

### 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 datasheets.

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

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 is submitted electronically will receive 100% verification prior to submission, 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.

1. invertebrate sampling involves the use of ethanol as a preservative. Ethanol is a toxic substance and should never be consumed or inhaled. Ingestion of ethanol can be fatal. Never store ethanol in an unmarked container, as it resembles water. Ethanol should be stored in a well-ventilated space, and fire code prohibits storing it in basements. Never pour ethanol onto the ground or into waterways. When disposing of ethanol, it should always first be diluted in a 4:1 ratio with tap water, for instance 4 liters of water for every liter of ethanol. Always wear gloves when handling ethanol. If you spill ethanol on your skin, immediately wash the affected area and alert your sampling partner(s). If you spill ethanol on clothing, immediately change the affected clothing. Before transporting ethanol, ensure the container is securely closed. Whenever ethanol is not in immediate use, close the container securely. Transporting ethanol also requires care. Containers with it should be carefully closed and secured to prevent rolling around or ejection during transit. The containers should be clearly marked. [↑](#footnote-ref-1)