

Monitoring and Sampling Manual

Environmental Protection (Water) Policy 2009

Prepared by: Water Quality and Investigation, Department of Environment and Science (DES)

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Refer to the individual sections of this document for the version history of each section.

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Foreword

The Queensland Government undertakes water monitoring for a number of reasons including: providing information to government for policy and investment decision-making, to underpin natural resource management decisions by government and stakeholders, to assess environmental impacts, and to educate and inform stakeholders and the community.

Monitoring is also conducted by local government, industry, regional NRM bodies and community groups. Many of these organisations collect valuable information on the condition of Queensland waters that complements Queensland Government monitoring.

The Monitoring and Sampling Manual 2018 is an update to the 2009 version and provides the common techniques, methods and standards for sample collection, handling, quality assurance and control, custodianship and data management for use by Government agencies, relevant persons and other organisations.

Where monitoring is required under legislation to be done under a protocol, the Monitoring and Sampling Manual 2018 is the principal document to decide the protocols. This manual is also intended to be used by persons and organisations involved in the monitoring of the condition and trend of Queensland waters.

The Monitoring and Sampling Manual 2018 will help ensure that monitoring data available to all stakeholders is consistent and scientifically accurate. The data will help users assess the condition of Queensland waters and trends in water health. This is aimed at ensuring the aquatic environment can be managed for sustainable development and aquatic ecosystem health.

Purpose

The purpose of the Monitoring and Sampling Manual 2018 is to provide the common techniques, methods and standards for sample collection, handling, quality assurance and control, custodianship and data management, for use by Queensland Government agencies, relevant persons and other organisations.

The Monitoring and Sampling Manual 2018 is a part of an integrated monitoring framework to decide the priorities, indicator selection, data storage, data analysis and reporting.

Where monitoring is required under legislation to be done under a protocol, including the Environmental Protection (Water) Policy 2009 and the Environmental Protection Regulation 2008, the Monitoring and Sampling Manual 2018 is the primary document to decide the protocols.

The Monitoring and Sampling Manual 2018 supersedes sampling manuals published by the former Environmental Protection Agency, Department of Primary Industries, Department of Natural Resources and Water, and Department of Environment, Natural Resources and Mines.

Section A: Sampling Design and Preparation

Sampling scope and design

Version: February 2018

1. Sampling scope and design

1.1. Purpose and scope

This document outlines the key elements that need to be taken into account when developing a sampling program. This document is adapted from the ANZECC and ARMCANZ (2000) *Australian Guidelines for Water Quality Monitoring and Reporting*. It should be read in conjunction with *Sampling design and preparation—Preparation for sampling* as logistical issues may influence sampling design (e.g. holding times).

1.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Preparation for sampling*
- *Choosing a laboratory and analytical method, holding times and preservation.*
- *Quality control for water and sediment sampling*
- *Control and reference sites*

Physical and chemical assessment: Background to event monitoring

1.3. Introduction

Some of the primary reasons for monitoring and assessment covered by this manual are:

- Investigating pollution incidents. In such cases, the aim of sampling should be to obtain evidence that will:
 - discover and prove the source, nature, extent of impact and the effects of the contaminants
 - be performed in such a way as to be legally admissible in court
- *Confirming compliance to licence conditions of an environmental authority or development approval or other statutory provision.* Key points are:
 - To test for compliance with the licence conditions, samples must be collected in a manner that will ensure valid analysis results for those particular contaminants. Reference should be made to the current conditions of any relevant licence or permits. The conditions may include specific sampling locations, times of release and water quality characteristics that will assist with designing the sampling strategy.
 - Where the aim of the sampling is to measure compliance with conditions of an environmental authority or development approval, and the conditions include a statistically robust sampling regime, this should be followed. However, if there is reason to believe variability is a confounding factor, additional samples may be necessary for further investigation.
 - Where the statutory provision is not explicit, the sample should fairly represent the body of material from which it is taken during the period of the sampling.
- *Undertaking a receiving environment monitoring program.*
- *Undertaking an environmental evaluation of an activity.*
 - The Environmental Protection Act 1994 and its subordinate legislation, including the *Environmental Protection Regulation 2008* and the *Environmental Protection (Water) Policy 2009*, must be taken into account when deciding where and when to sample for a pollution investigation, checking compliance with an environmental authority or development approval, or undertaking a receiving environment monitoring program.

Before any sampling is carried out, the objectives aim of the sampling exercise, and how the results will be used should be established (scope of sampling). That information will help identify where and when sampling should take place, and the parameters that need to be determined for those samples (sampling design). Tasks associated with implementing the sampling design are presented in *Sampling design and preparation—Preparation for sampling*.

Essential features of a sampling strategy include ensuring that:

- samples collected are representative of the source material (i.e. waters, sediments and biota in a creek, river or lake) at the location of interest
- variation is taken into account – both in space (spatially) and over time (temporally)
- in situ measurements are reliable
- the integrity of materials sent for laboratory analysis has not been compromised by contamination, degradation or transformation
- sufficient sample volume is meet required detection limits for a particular analytical method and appropriate collection methods are used
- reference or control sample data are collected
- consideration of flow conditions (whether event or ambient) (See Physical and chemical assessment—Background to event monitoring).

Sampling designs should ultimately be defined by program objectives that can include the required statistical power required for discriminating between hypotheses or be based on the levels of acceptable sampling variability.

1.4. Importance of understanding the system being sampled

An understanding of the ecosystem is important to achieve a good sampling design. This understanding is best formalised in a conceptual model (or process model) of the system being examined. The model can be a simple box diagram that illustrates the components and linkages in the system, or a graphical representation of the system. Whatever model is used, it should present the factors that are influencing the system and the linkages of these factors.

During the formulation of a model, several decisions must be made or the model will be too complex. For example:

- What are the major issues of concern (e.g. nutrients, metal loads, bioavailable metals)?
- What ecosystem (including subsystem type) should the model describe (e.g. freshwater, marine waters, estuarine waters, wetland, seagrass bed, mangroves)?
- Which state of flow should the model describe (e.g. base flow, flood event)?

Once formulated, the process model can be used to help define:

- important components of the system and the important linkages
- key processes
- cause–effect relationships
- important questions to be addressed
- spatial boundaries
- valid measurement parameters for the processes of concern; what to measure, and with what precision
- site selection
- time and seasonal considerations.

An example of graphical conceptual model that may assist sampling design is presented in **Figure 1**.



Figure 1: Conceptual diagram of a coastal system including anthropogenic activities, inputs to waterways and areas of value

1.5. Why Sample?

The objectives of the sampling program should be determined and documented. These should be as specific as possible. Common sampling objectives include:

- determining if one or more contaminants found in the environment have originated from one or more sources
- determining whether one or more contaminants in a release are in sufficient quantity to cause adverse environmental effects consistent with those observed at the time of the incident
- determining whether the contaminants in a waste release are having a measurable impact on the receiving environment water quality and whether environmental values are being affected
- determining whether the quality of waters have changed significantly, consistent with the definition of the term 'environmental harm' in the Environmental Protection Act 1994 as a result of a release.

When sampling the receiving environment your assessment should take into account:

- potential sources of contamination/releases
- likely contaminants
- type of waterway and flow rates, whether:
 - freshwater, estuarine or marine
 - a flowing stream, lake, or ephemeral/temporary waters (in which case it may be wet or dry, or evaporating and concentrating contaminants at the time of sampling)
- licensed releases into the waters
- recent weather such as heavy rain, showers or drought conditions
- historical occurrences of similar incidents.

1.6. Spatial boundaries of sampling

The geographic boundaries of the sampling event should be based on the issue of concern and the ecosystem type rather than on convenience and/or budgets. For example, some important considerations would include:

- the likely spatial uniformity of the parameter/s of interest at a location (e.g. at depth, cross section of a river)
- the extent of the potential impacts downstream.

It is important to ensure that the sampling regime is representative of the system and parameter/s of interest. For example, where a water body is well mixed and a parameter of interest is evenly distributed in the water column, a grab sample may be appropriate. However, if water quality changes with depth, a number of samples at different depths may be required.

1.7. What to sample

Environmental authority or development approval conditions typically specify the contaminants and the permitted ranges of concentrations allowed in the release. However, in cases of suspected environmental pollution incidents, the pollutants present may be unknown and an assessment should be made on potential sources of contaminants in the area.

When sampling the receiving environment, there may be a range of related indicators that will be measured across different media such as surface water, sediments or biota. Indicators are physical, chemical or biological measures that best represent the key elements of an ecosystem. When choosing indicators, it is important to know whether there are defined benchmarks such as water quality objectives, guidelines, limits or other standards to compare measured data to. Indicators may be chosen because they have such benchmarks and may best indicate water condition or potential environmental harm. Sources of guidelines and benchmarks for Queensland are:

- Water Quality Objectives and Environmental Values prescribed under the Environmental Protection Policy (Water). Available from: <http://www.ehp.qld.gov.au/water/policy/index.html>
- DEHP (Department of Environment and Heritage Protection) 2009, Queensland Water Quality Guidelines, Version 3, ISBN 978-0-9806986-0-2. Queensland. Available from: <https://www.ehp.qld.gov.au/water/pdf/water-quality-guidelines.pdf>
- ANZECC and ARMCANZ (Australian and New Zealand Environment and Conservation Council and Agriculture and Resource Management Council of Australia and New Zealand) 2000, Australian and New Zealand Guidelines for Fresh and Marine Water Quality, National Water Quality Management Strategy, Paper 4. ANZECC and ARMCANZ, Canberra. Available from: <http://www.environment.gov.au/system/files/resources/53cda9ea-7ec2-49d4-af29-d1dde09e96ef/files/nwqms-guidelines-4-vol1.pdf>
- NHMRC (National Resource Management Ministerial Council) 2008, Guidelines for managing risks in recreational water. Australian Government, Canberra, 216p. Available from: http://www.nhmrc.gov.au/_files_nhmrc/publications/attachments/eh38.pdf
- NHMRC and NRMMC (National Health and Medical Research Council and National Resource Management Ministerial Council) 2011, Australian Drinking Water Guidelines, National Water Quality Management Strategy, Paper 6. NHMRC and NRMMC, Commonwealth of Australia, Canberra. Available from: www.nhmrc.gov.au/guidelines/publications/eh52

Under the National Water Quality Management Strategy (ANZECC & ARMCANZ 2000) water quality framework and the Environmental Protection Policy (Water), properly developed and approved local guidelines hold higher precedence over state or national guidelines. State (or national) guidelines apply when local guidelines do not exist, and are used for any parameters that are not included in the most locally relevant guidelines.

Although environmental authority or development approval conditions typically specify contaminants and the permitted ranges of concentrations allowed, additional characteristics may provide greater information about the potential environmental harm that might be caused. For example, although only biochemical oxygen demand (BOD) might be specified in the environmental authority or development approval, chemical oxygen demand (COD) and total organic carbon (TOC) often provide more information, and may be worth assessing. It may also be important to measure other characteristics due to a change in an operating condition or a specific incident.

In addition to measuring water quality characteristics, flow measurement of wastes/wastewater is often required for point source releases. This allows regulation and quantification of flow and loads of contaminants. Flow measurements of water bodies can be important for regulation as they can be used to assess initial mixing of point source discharges or as triggers that permit the release of licensed discharges, particularly for event-based releases. Flow measurements of waterways may also be required for pollution incidents to assess or predict the extent of impact. Any monitoring for these purposes should be in accordance with National Industry Guidelines for hydrometric monitoring (<http://www.bom.gov.au/water/standards/niGuidelinesHyd.shtml>).

1.8. Where to sample

Many environmental authorities have conditions that specify where samples are to be taken. Some have more than one sample point (two or more outlets, or an intake as well as an outlet). Where no sampling location is specified, samples should be collected from a site representative of the release material (and the receiving waters, where relevant).

When investigating environmental pollution incidents, consider all possible sources of the pollutant (including licensed and unlicensed sources of release). Samples should be collected:

- at the site of the reported pollution
- at the point of any contributing or suspected sources
- in an area upstream from the suspected source/s (control site)
- at points downstream of the suspected source (to measure extent). Samples should be collected as far downstream from the source as suspected of being polluted.

Reference or control sites must be sampled (if water is present) in order to understand the background conditions at the time of sampling, and in order to fully understand the potential impact from the pollution event under investigation. If assessing sediment, reference or control sites must be sampled. See Sampling design and preparation—Control and Reference sites.

It is important to identify with sufficient accuracy the location from which a sample has been collected to avoid raising a doubt about 'what the sample represents', particularly in cases where a location a few metres away might have given significantly different results. This is done by using a GPS and making a mud map in notes about exactly where a sample was collected from. See Sampling Design and Preparation— Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection.

Water bodies are not homogeneous within a cross sectional area or depth profile and can be stratified (layered). This means that the composition of the different layers is substantially different in respect of at least one characteristic. For example, in estuaries, water quality characteristics can vary because of ingress/egress of saline waters. Estuaries are commonly stratified when freshwater flow is much larger than tidal flow—the fresh flows seawards over the saline waters and a 'salt wedge' develops. Stratification can also result from temperature effects in waters with low current velocities (such as lake, dams and pools). Such stratification is usually most pronounced in summer months when surface waters are much warmer than bottom waters. After separation, the water layers often develop markedly different chemistry. Such layers also tend to prevent mixing of discharged contaminants. The reverse process (de-stratification) can occur when the seasons change. The resulting inversion ('turnover') of the water can result in low oxygen water rising to the surface and causing adverse effects (such as odours from anaerobic decomposition at depth, and/or nutrient/metal enrichment). Other examples include the distribution of suspended solids within the water column from physical processes of re-suspension, deposition and flocculation.

When sampling environmental waters (typically, when investigating a pollution incident), it is important to remember that stratification might have occurred. In situ measurements taken at different depths can be used to detect stratification and sampling can be adjusted accordingly.

1.9. When to sample

Virtually all waters show both temporal and spatial variations in quality and so the timing and choice of location/s for taking water samples must be chosen with care. The quality of sediments and biota will also vary over time and space, although these changes may occur over a longer period than those detectable in water samples.

If the variation within a water body is not understood, it may be necessary to establish a pilot sampling program to determine the variability and determine the optimum sampling program. In circumstances where undertaking an assessment of variability is not practical or possible, it is recommended that information from relevant peer reviewed literature on the likely variability is used to provide guidance on an appropriate sampling strategy. A similar approach (for dealing with inherent variability) is recommended when designing sampling programs involving the collection of other materials such as sediments and biota.

The schedule for the sampling program should take account of the expected temporal resolution of changes in the environment. For example, in programs for monitoring wastewater treatment effluents, sampling around the clock may be required to determine whether control variables have been met or exceeded.

In terms of frequency, sampling may be required every hour, day, week, fortnight, month or possibly only once a year. The frequency of sampling (level of resolution) should be sufficient to meet the requirements of the program objective but not cost more than necessary. Composite sampling and passive samplers can be used to integrate variations in water quality over an extended period of time.

Some conditions on environmental authorities specify that release is to take place only at certain times of the day (for example, on an outgoing tide) or under certain weather conditions. This should be considered in your sampling

design where applicable.

For impact assessments, sampling before and after is important (but not always possible), preferably with multiple before and after reference sites. In situations where there is no 'before' information available at the impact location, data collected by sampling from reference or control sites may be indicative of conditions at the impact location prior to the incident. For example, a chemical spill may have contaminated the receiving environment, and caused impacts on local biota, but there are no pre-spill data available. However, concentrations of contaminants or biological community indices measured at unimpacted reference or control sites after a chemical spill can be indicative of what those parameters could have been at the incident location prior to the spill.

Water quality varies with stream flow conditions, so when considering the timing of sampling, it is important to establish whether sampling during baseflow or during flood event conditions (or both) is appropriate. See Physical and chemical assessment—Background to event monitoring for more information.

1.9.1. How many samples should be collected?

Unless the material being sampled is known to be well mixed (well mixed water body or end-of-pipe discharge), it is unlikely that a single measure will be representative of the source body of material. Multiple measurements are needed to allow the calculation of descriptive statistics (i.e. a mean and confidence interval, or percentile statistics) for the characteristic of interest, or to allow statistical testing for significant differences between locations or non-compliance with statutory provisions. This requires multiple readings for in situ measurements and multiple samples where laboratory analysis is involved. A minimum is three data points per site for basic statistical tests, but more may be required depending on the inherent variability in the measurement data. The number of data points needed may not be known until after chemical analysis of some samples. It is good practice to take additional samples and to store these for subsequent analysis if required, although short maximum holding times for some contaminants may mean this is impractical.

1.9.2. Is grab sampling adequate or should composite samples be taken?

Most samples taken will be grab samples—taken by filling sample containers over a 'short' period (seconds or minutes). A single grab sample may be used where:

- a hazardous situation has arisen or is suspected and the sample is taken to confirm the presence of the hazardous substance
- where the body of water being tested is well mixed and its quality can be adequately described by a single sample.

Note: A single grab sample can be of limited use if it takes no account of variations in quality with time or space. In such a situation, the taking of a composite sample is a useful strategy. A composite sample may be:

- **temporal** by combining contributions of material collected over a longer period (minutes, hours or days).
- **spatial**, for example, comprising a series of equal contributions of material taken along a transect (e.g. across a channel). This gives a spatially 'more representative' sample than a single grab sample at a single point.

However, composite sampling will not provide information on the maximum concentration in a series of samples, and will provide an average. Depending upon the situation, the maximum concentrations can be important when dealing with toxicants¹.

1.10. Quality Control

Quality control is an essential component of any sampling exercise. The purpose of a quality control scheme is to check whether bias, sample contamination, or analyte loss could affect the results, and so invalidate the process. Quality control is discussed in more detail in Sampling Design and Preparation: Quality control for water and sediment sampling.

¹ A toxicant is a chemical capable of producing an adverse response (effect) in a biological system, seriously injuring structure or function or producing death. Examples include pesticides, heavy metals and biotoxins (i.e. domoic acid, ciguatoxin) (ANZECC and ARMCANZ 2000)

1.11. Cost effectiveness

It is preferable for the cost of sampling programs to be as small as possible while still meeting the stated objectives of the monitoring study. Cost-effectiveness considerations involve trade-offs between loss of statistical 'power' (i.e. the capacity of a program to discriminate between various hypotheses) and the cost of data acquisition. Costs of data acquisition taken into account for cost effectiveness include:

the number of sampling stations, sampling occasions and replicates

- the cost of collecting samples (staff, transport, consumables)
- the cost of analysis
- the cost of data handling and interpretation (cost of reporting).

Cost-savings can result from collaborative monitoring, for example, when local councils pool resources with other water managers to comprehensively monitor a particular water body.

1.12. References and additional reading

ANZECC and ARMCANZ (Australian and New Zealand Environment and Conservation Council and Agriculture and Resource Management Council of Australia and New Zealand) 2000, Australian Guidelines for Water Quality Monitoring and Reporting. National Water Quality Management Strategy. No. 7. Available from: <http://www.agriculture.gov.au/SiteCollectionDocuments/water/nwqms-monitoring-reporting.pdf>

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NHMRC and NRMCC (National Health and Medical Research Council and National Resource Management Ministerial Council) 2011, Australian Drinking Water Guidelines, National Water Quality Management Strategy, Paper 6. NHMRC and NRMCC, Commonwealth of Australia, Canberra. Available from: www.nhmrc.gov.au/guidelines/publications/eh52

Water Quality Objectives and Environmental Values prescribed under the Environmental Protection Policy (Water). Available from: <http://www.ehp.qld.gov.au/water/policy/index.html>

Preparation for sampling

Version: February 2018

2. Preparation for sampling

2.1. Purpose and scope

This document outlines all aspects of preparation for water and sediment sampling. It also outlines secure handling of samples from the point of collection to the laboratory or court.

2.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Choosing a laboratory and analytical method, holding times and preservation*
- *Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection*

Physical and chemical assessment: Water quality sampling using in situ water quality instruments.

2.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

2.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

2.5. Skills, training and experience

When preparing for field work, ensure staff are available who have the necessary skills to undertake the methods that will be used in the sampling program. Also consider whether site specific inductions are required.

2.6. Things to consider when preparing for field work

2.6.1. Preparation of a sampling schedule

A sampling schedule should document:

- whether permits or approvals to enter and sample land are needed
- a landholder communication plan
- review of any previous work
- a site plan which identifies specific locations where the samples are to be collected
- when the samples are to be collected
- how the samples will be collected (sampling protocols)
- types of samples to be collected (including quality control samples)
- who will be doing the sampling and skills/training required
- analysis required for the samples and what laboratory will be undertaking the analysis
- sampling containers and associated equipment needed
- preservatives and sample storage conditions needed
- how the samples will be referenced and recorded during collection
- the maximum holding time for each sample
- health and safety considerations
- chain of custody (COC) and other evidentiary requirements
- equipment cleaning/decontamination method
- transport logistics (e.g. if remote consider distance/time from sampling location to courier and laboratory etc.)
- other records or information that may need to be obtained.

2.6.2. Naming of sites and samples

Where possible, it is best to decide on names or a naming convention for the sites and samples before heading out to into the field. Calling a site 'Site A' is not sufficient – the name should be unique to the site and the project. At each site, a number of samples may be collected – for example, a water sample and a sediment sample, or replicates. To avoid confusion, each sample must be given a unique name/identifier, such as PROJECT-SITE-SED for sediment (e.g. TEXAS-DAM1-SED) and PROJECT-SITE-WAT for water. A number of containers may be used to collect each 'sample'. These containers must have the same sample name recorded on each container. The number of containers per sample must be recorded. The sample identifier (Sample ID) needs to be as short as possible and individual, but also contain sufficient information to describe the sample properly (for example project name, sample location, sample type, and sample depth). Keep in mind the ID needs to be able to fit on a sample jar label or a sample bottle label. Also, for sediment samples, the depth is an important inclusion because several samples are often collected from the same location. Sample date and time, sampler, etc. must also be recorded on the sample jar and the COC. If a site is a routine monitoring site, a consistent name should be used for easy data review. For example, site names documented in an Environmental Authority (EA) should be given to the same sites sampled under an associated Receiving Environment Monitoring Program (REMP).

2.6.3. The Sample Register

If multiple people are collecting samples for an investigation, a person should be nominated as the official property officer prior to attending site. All samples should be handed to this person who takes custody of all the exhibit/samples, and must be able to prove at all times the whereabouts of these items when in their control. Where samples are taken away or delivered to a third party (for example a laboratory) this person needs to record and log the removal from their custody and hand over to another party. This can be signed for and receipted in the sample register. The sample register must contain the following information for each sample:

- The unique name / identifying number: e.g. TEXAS-DAM1-SED01
- Description: e.g. Security-sealed plastic bag containing a plastic bottle with soil samples.
- Time the sample was taken: e.g. 2.45pm
- Where taken: e.g. GPS location and/or river bank LHS (very important that the exact location is identified)
- By whom: e.g. Jane Smith DES

The sample register then needs to have a declaration as to the transfer / handover of possession. For example, "At (time) and (date) the samples attached to this register were handed into the possession of (name – position)..." or "... At (time) and (date) the following samples were handed into the possession of (name – position)". This would usually be at the bottom of the register. This is a different document to the chain of custody (COC) document required by laboratories.

This register forms the contemporaneous notes taken at the time and will form the basis of any evidence you may later be asked to produce in court.

2.6.4. Communicating with the laboratory

2.6.4.1. Before sampling

Before undertaking sampling, a laboratory needs to be chosen to undertake the analysis of the samples. See Choosing a laboratory and analytical method, holding times and preservation document for more information.

It is important to contact the analytical laboratory before going into the field (if possible). Laboratories differ in what they can analyse, and their requirements (such as sample volume and preservatives). Points to discuss with the laboratory include:

- the analysis required and whether the laboratory can reach the required limits of reporting (LOR)
- the procedure for ordering sample bottles (and whether they can be individually sealed in plastic 'tamper proof' bags if being used in an investigation) (**Figure 2**)
- the delivery time for any sample containers ordered
- the laboratory's recommended preservation requirements for the analyte of interest
- the laboratory's maximum holding times for the analyte of interest
- the quantity of samples to be submitted
- sample submission and chain of custody (COC) documentation
- the date and time of sample submission to the laboratory
- the turnaround time of the analysis
- conditions under which re-analysis can be conducted.

When in the field, communicate with the laboratory about any changes from the original plan that may occur

(particularly in relation to when the samples will arrive).

It may also be useful to inform the laboratory of:

- the sample source
- the likely range of concentrations
- the purpose for which the results are to be used, or if high concentrations are expected (e.g. if the sample is from an area affected by a chemical spill).

The benefits of providing the laboratory with as much information as possible include:

- assisting the analyst in choosing a suitable analytical method with an appropriate LOR. In some cases the LOR can be improved if the analyst knows these details beforehand
- avoiding subsequent delays because samples need to be diluted and re-analysed.



Figure 2: Sample containers security-sealed in plastic

2.6.4.2. After sampling

The receiving laboratory should be notified in advance that samples are to be dispatched. An acknowledgment of notification should be received back from the laboratory prior to sending the samples to them. Only in exceptional circumstances should samples be sent without prior notification. Samples delivered to the laboratory must be handed to a supervisor or appropriate responsible staff member. This person should acknowledge receipt of the samples by signing the consignment documents accompanying each sample carrier box, chain of custody documentation or other appropriate form of receipt.

The analyst must contact the sampler promptly if there are any signs of tampering or other irregularity. In such cases, the sampler and analyst should discuss and decide whether analysis should proceed or whether re-sampling is required. Record the details of this discussion.

Note: The analyst's report should include details of the condition of the samples on receipt. If any irregularity was discovered on receiving the samples, the analyst must also give details of this.

2.6.5. Anticipating potential court action

Samples and their method of collection may be required to be proven in a court of law at a later date. Each part of the collection process, the transportation to the laboratory and the subsequent hand over needs to be clearly explained and documented (including COC documentation). If samples are required for a court action, failure to do this may render any results inadmissible in court.

2.6.6. Sample containers, preservation, storage and holding times

2.6.6.1. Sample containers

Sample containers and preservation techniques may vary from laboratory to laboratory (in accordance with which Australian and international standard methods they are using), and so it is necessary to discuss the requirements with the laboratory prior to ordering containers. Samples should be collected into the appropriate container and stored as per the requirements of the analysing laboratory. Sample containers should have a water proof label

attached with space for the sampler to fill in appropriate details.

Note: If investigating a pollution incident, the situation may arise where sampling is required even though the correct sample container is not available. As there is often only one opportunity to sample in this situation, do not automatically assume that it is not possible to sample. A level of cautious improvisation should be considered such as using alternative clean containers. Contact the laboratory for options.

2.6.6.2. Preservation, storage, and holding times

Before going into the field, ensure the preservation requirements and the maximum holding times for different parameters are known and can be met. Discuss holding time requirements with your analysing laboratory. The samples need to be delivered before the maximum holding times, with enough time for the laboratory to analyse the samples.

If preservatives are to be taken into the field, they should preferably be supplied in small vials sealed in plastic bags. The vials should be labelled with the following information:

- preservative type and quantity
- preservative expiry date
- batch number
- hazard warnings if necessary.

Clear labelling of all chemicals is essential and material safety data sheets (MSDS) should accompany chemicals at all times.

Some sample containers are supplied with preservatives in situ. This may be in the form of a small volume of liquid or crystals inside the empty container.

Samples should generally be stored refrigerated or frozen. Refrigerated samples should be kept at between 1 and 4°C (AS/NZS 5667.1:1998) for chemical samples, and at $5 \pm 3^\circ\text{C}$ for microbial samples (AS/NZS 2031:2012). The sample should be cooled to the correct temperature range as rapidly as is reasonably practicable and kept within that temperature range until analysis commences. When transporting samples, they should be transported in a chilled or frozen state in line with storage conditions (see sections 2.6.7.1.1 and 2.6.7.1.2).

2.6.7. Transportation of samples

The transportation of samples needs to be organised in advance, to ensure samples arrive at the laboratory on time, in the condition specified by the laboratory and with the chain of custody intact. If using a commercial courier, discuss the time frames involved in the delivery, to ensure samples will arrive before the recommended maximum holding times have expired. Regularly check the status of the samples to ensure they do not get 'held up' somewhere.

When preparing for sampling, make sure any cooler boxes or refrigeration equipment have been cleaned thoroughly with appropriate cleaning equipment. For example, if a cooler box has been used for storing fish, and is then used for storing samples collected for nitrogen or phosphorus, residual odorous substances from the fish (such as ammonia) can permeate the container walls, even if the container is of high density polyethylene (HDPE).

Transportation options can include:

- personal delivery
- being sent via a commercial carrier (such as road transport or air cargo).

Note: Not all commercial transportation companies will ship all chemicals. Contact the courier company for details prior to sampling. Commercial carriers have shipping regulations – ensure the sample packaging and labelling meet the requirements. Samples sent by air are subject to the International Air Transport Association (IATA) Dangerous Goods Regulations (updated annually). Failure to comply with regulations can lead to prosecution of the consignor. Consult the airline company or the Civil Aviation Safety Authority (CASA) before sending samples to ensure the sample packaging (including the carrier box) and the labelling of the carrier box meet the requirements.

2.6.7.1. Packing samples

Samples need to be packed in a way that minimises the risk of breakage, leakage or spillage during transport. Some points to remember:

- Sample containers should be packed in an upright position so they do not fall over and potentially leak. Therefore, any extra space should be packed with plastic sheets or other inert material to keep samples upright.
- If undertaking ultra-trace sampling, it may be appropriate to double bag samples to protect them from cross

contamination from leakages and melted ice.

- Glass bottles and jars should be packed in bubble wrap.
- Freight should be labelled as fragile if containing glass jars/bottles, and use 'keep chilled' stickers if required.

2.6.7.1.1. Samples requiring refrigeration

In order to quickly cool samples when they are first taken, either place samples in a pre-chilled portable fridge, or place in a cooler box with crushed ice packed closely around the samples (use double bags if necessary). Points to remember:

- Do not place ice over the top of the sample containers as melted ice can potentially cause contamination.
- Keep the sample container lids above the level of the ice.
- If using ice to chill the samples, repack into a cooler box with frozen ice bricks once samples are cooled (approximately two hours). This prevents any risk of the samples being contaminated by melted ice.

2.6.7.1.2. Samples requiring freezing immediately after collection

For samples that require freezing immediately after collection, a pre-chilled portable freezer is the best option for sample storage. If this is not possible, an alternative is dry ice. Points to remember when using dry ice:

It is available in block and pellet form. Pelletised dry ice is preferable as it can be packed in much closer contact with the samples.

A combination of block and pellets can also be used, the pellets being placed next to the sample containers.

Suppliers of dry ice are listed in the telephone directory or can be found via web search.

Note: It is hazardous to transport dry ice inside a motor vehicle with all of the windows closed.

If a freezer or dry ice is not available, samples can be frozen by surrounding them with a slurry of crushed ice mixed with common domestic salt (sodium chloride). This rapidly achieves temperatures well below 0°C.

2.6.7.2. Chain of custody (COC) documents

Chain of custody (COC) documents record information about a sample/s including date, time, sample identification (ID), sample matrix, preservation type, and analyses required. Most importantly they provide a record who has had custody of the samples from field sampling through to the submission at the laboratory. The COC document should be signed each time a person hands the sample to another person, and should include the full name and title of the person receiving the sample. Each laboratory has their own COC documentation which should be acquired prior to sampling.

2.6.8. Security of samples

If samples are to be used for legal proceedings, it must be demonstrated that there was minimal risk of interference with the samples between the time of sampling and the time of analysis, hence the nomination of a property officer and the use of the sample register. This requires a well-designed system for security of the samples, including precautions to make any such interference evident upon receipt by the analyst. The date and times of transfer and to whom, need to be recorded. The security of samples is particularly important if samples are to be sent using a courier company (and not a direct handover from the sampler to the analyst/laboratory). Ensure the laboratory receiving the samples is aware that the samples may be required for legal proceedings and that they are handled appropriately. The more times samples are handled the greater the risks and issues surrounding the chain of custody. Hence, there is a need to minimise handling with third parties. Make sure that any third party details are recorded in case they need to provide evidence of their handling of the samples in court e.g. name, address and contact details.

2.6.8.1. Sample seals or evidence bags

Either sample seals (**Figure 3**) or evidence bags can be used to secure samples. Typically seals are specially printed self-adhesive 'security' labels, designed to be affixed across the body and cap of the sample container. Each seal is made of a 'self-destruct' material so that any attempt to remove it will result in its disintegration and cannot be re-affixed in its original condition.

Include the seal/evidence bag number and the sample identification number when recording details of samples.



Figure 3: Example of security labels and seals

2.6.8.2. Locked carrier boxes

One way to hinder unauthorised access to samples is to use a system of insulated carrier boxes fitted with locks that can be opened only by:

- an appropriate staff member of the organisation with the authority to do so
- the analyst or other laboratory staff member having similar authority.

The two parts of the assembly need to be fastened by (for example) suitable rivets, rather than screws, as screws can be removed and replaced without leaving evidence of the fact. Locks should:

- be fitted in a way that ensures the lock cannot be removed without leaving evidence of tampering
- be part of the body of the carrier box, or a padlock that fastens a hasp and staple assembly permanently fitted to the body; and that they are
- case hardened to resist cutting by a hacksaw.

Note: If this system is used, it should be possible to testify in court that the keys were kept in secure places.

2.7. Preparation of equipment

Equipment needs to be checked to make sure it is in good working order, to avoid breakages and delays whilst in the field. All equipment that will be used for sampling needs to be cleaned and prepared before going into the field. This is to ensure there is no chance of contamination from previous sampling. Any equipment that requires calibrating must be calibrated to the manufacturer's specifications before and checked after sampling. Records of all calibrations need to be kept. See Water quality sampling using in situ water quality instruments.

2.8. Essential items

The following section provides some items considered essential for sampling in the field. They are also summarised in a checklist in Appendix 1.

- Basic health and safety items:
 - call in/emergency procedure
 - personal protective equipment (PPE) such as life jacket, zinc free sunscreen, hat, appropriate clothing, insect repellent
 - drinking water
- communication devices (such as phones, satellite phones, SPOT devices).
- Sampling schedule
- Sample register
- Contact list:
 - all field staff on the sampling trip
 - office staff who may need to be contacted
 - landholders who have given approval to enter their properties
 - analytical laboratory contact details to inform them of any change in plans or to ask questions
 - transport companies.
- Global positioning system (GPS) (see Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection document)
- Camera and/or video camera/body cam – photos can be considered evidence, and therefore, need to be stored in a secure location. Be prepared to immediately download digital images on return to the office
- Voice recorder – a voice recorder can be useful for immediate recording of your observations if circumstances

make writing on paper difficult. If you use one, you should listen to the recording and transcribe it as soon as practicable

- Maps – maps or aerial photographs showing the location where sampling is to be undertaken should be taken to site. The maps should also indicate the best route to the sampling site, if it is private property and any other potential sampling sites
- Smartphone/tablet - with maps and other applicable information
- Marking pens – only waterproof pens should be used for labelling samples (enamel paint pens are useful). When sampling waters for the presence of solvent-type compounds, extra caution should be used because marking pens contain solvents and could contaminate the sample (e.g. look for a xylene free permanent marker)
- Notebooks, field sheets, pens and pencils
- Methods for sampling
- Chain of Custody (COC) paperwork and other documentation required by the laboratory/transport company
- All equipment associated with the specific method to be used in the field
- Spare equipment – pack spare batteries, pens, markers, sampling equipment and containers where possible. This is to allow for breakages, lost equipment and extra ad hoc sampling.

2.9. References and additional reading

AS/NZS 2031:2012, Selection of containers and preservation of water samples for microbiological analysis.

AS/NZS 5667.1:1998, Water Quality—Sampling—Guidance on the design of sampling programs, sampling techniques and the preservation and handling of samples.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Personal protective equipment (PPE) such as life jacket, zinc free sunscreen, hat, appropriate clothing, insect repellent	
Drinking water	
Communication devices	
Sampling schedule	
Contact list	
GPS	
Sample register	
Evidence bags, seals	
Camera/video/voice recorder	
Maps	
Marking pens	
Notebooks, field sheets, pens and pencils	
COC and other laboratory and transport paperwork	
All equipment associated with field methods (e.g. water sampling, sediment sampling) and spare equipment	

Record keeping, including taking field photographs and videos

Version: February 2018

3. Record keeping, including taking field photographs and videos

3.1. Purpose and scope

This document is a guide to ensure accurate record keeping in the field. Accurate and complete record keeping of site and sampling information is vital for completing field surveys and investigations. Information collected in the field, including notes, voice recordings, photographs and videos, may later be used as evidence in a court. They also support and corroborate the witness's observations and actions on the day.

3.2. Associated documents

Sampling design and preparation: Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection

3.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

3.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

3.5. Skills, training and experience

No skills, training or experience is required to conduct the procedure within this section.

3.6. Equipment

- Equipment specific to this method includes:
- camera
- video recorder/Go Pro or body cam
- GPS
- voice recorder
- official notebook, field sheets or other method of recording
- a ruler
- pens, markers, pencils etc.

3.7. Procedure

3.7.1. Prior to undertaking fieldwork

- 1.1. Ensure the digital equipment (e.g. camera, video recorder, voice recorder, GPS) are fully charged and spare batteries are packed.
- 1.2. Check the date and time on the digital equipment against your computer prior to undertaking the field survey. If incorrect, follow the equipment manual to update the date and time on your digital equipment.
- 1.3. It is recommended that the numbering of the images on the camera/video recorder is reset to zero.
- 1.4. Ensure there is an accurate method for record keeping (i.e. field sheets, notebook) suitable for the investigation. If the results are likely to be used in court, the information should be written into an official notebook (i.e. bound spine with consecutively numbered pages to show no pages have been removed) or an electronic log alternative. A digital voice recorder can be used to assist in record keeping, however details of on-time and off-time still need to be recorded elsewhere.

- 1.5. Make a record of the digital equipment to be used during the field work (e.g. model, make and year of manufacture).
- 1.6. Calibrate the GPS unit (see document Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection).

Note: It can be useful to pre-fill some standard information such as date, investigation name, and other site details if known.

3.7.2. Undertaking fieldwork

- 1 Upon arrival at the site, record the time in notebook or field sheet.
- 2 Record the full names, position and roles of persons present.
- 3 Ensure that you have a plan with respect to the roles and duties of each person in the field team. For example, Person 1 is the overall controller of the site, Person 2 is designated the role of filming/recorder, Person 3 is collecting samples etc. Where practical, identify a property officer whose primary responsibility is the recording, collection and continuity of all seized samples.
- 4 Record general site information relevant to the investigation such as weather conditions, vegetation disturbance, rubbish, scums etc.
- 5 Record the GPS location using the method described in document Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection. If storing positions as waypoints, note the waypoint number in notebook/on field sheets while in the field.
- 6 Nominate one person to take photographs of the site.
- 7 Whilst still standing where the GPS point was taken, take initial photographs of the site:
 - For terrestrial environments, take eight photographs in eight directions (N, NE, E, SE, S, SW, W, NW) so that photographs all have side-lap and can be joined in a panorama of the site. Include a hat/person/bag in the first photo (north) and always rotate in a clockwise direction looking down at your point.
 - For aquatic environments, take four photographs in four directions (downstream, right shore, upstream, left shore), rotating in a clockwise direction.

These photographs are taken as a backup to the GPS point in case of GPS error.

- 8 Record each photo number and time taken in the notebook/field sheets, making additional notes of what the images depict.
- 9 Take any additional photographs/videos relevant to the investigation, recording the time, location, photo number and a description. It is essential to clearly identify when and where each shot is taken. If possible, include an identifying feature in the photograph (e.g. site label) to assist when examining photographs at a later date. The use of a scale (i.e. ruler, pen) is useful for close-up shots.
- 10 Photograph all samples as they are collected and make a record of the samples as they are collected.
- 11 Where practical a video (Go Pro or body cam) / camera should be used for the identification and recording of samples being collected notwithstanding the notes taken at the time.
- 12 Record any measurements taken at the site (if relevant).
- 13 Record any relevant conversations with landowners, general public, or company representatives.

Note: if using a voice recorder, advise the people involved prior to recording that the conversation is being recorded. It is an offence under the Invasion of Privacy Act 1971 to use a voice recorder to record private conversations where the person recording the conversation is not a party to the conversation.

- 14 Complete a site sketch, providing an overview of the site area (with indication of direction – North), location of initial photographs and GPS location, location of sample collection point/s and any other information relevant to the investigation.
- 15 Prior to leaving the site, review notes to ensure all relevant information has been recorded and fill in any gaps that may be present.
- 16 Repeat steps for each new site.

Note:

- If taking close up photographs, it is advisable to use a ruler as a scale for the photograph.
- If undertaking an investigation, consider the use of plastic alpha-numeric evidence stands, markers or flags to identify the exact location of sampling points in a terrestrial environment, or where evidence is found at a site. Establishing a reference point with a marker, recording GPS co-ordinates at each marker and generating unique identifier for each sample provides for accurate recording of exactly where samples were taken enables easy cross referencing onto a map. The area should be photographed in situ from all angles prior to sampling and again photographed/digitally recorded as markers are placed at the various sampling points.
- It is important when taking photographs to accurately determine the location of each photograph. This may be by using a camera with an inbuilt GPS, including a permanent land mark in the photograph or by marking a tree

with a permanent mark (e.g. spray paint) if appropriate.

- Ensure note taking is neat, precise and contains only the facts (who, what, when, where, why, how).
- A clear record should be made of all photographs, videos, measurements and samples, including time, location and descriptions.
- If using a voice recorder, record the start and finish time in the notebook/field sheets. When the voice recorder is turned on, state the date, time, location and persons present.

3.7.3. Upon return to the office

2. Check and record the accuracy of the date and time on the digital equipment against your computer upon return from the field survey.
3. Download all photographs, videos, voice recordings and GPS co-ordinates (see document Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection). If the purpose of the field work was for an investigation or there is a possibility of it going to court, an original set of this data must be downloaded onto a CD/DVD, or similar non-rewritable device. No deleting, renaming or editing the files in any way can occur prior to doing this.
4. Scan the original notebook or field sheets.
5. Store downloaded information in an appropriate location. For investigations or legal cases, this information must be stored in a secure, locked location.
6. Store and maintain digital equipment in accordance with manufacturer's instructions.

Quality control for water and sediment sampling

Version: February 2018

4. Quality control for water and sediment sampling

4.1. Purpose and scope

This document outlines an approach for collecting control samples during a monitoring program—it does not cover laboratory quality control procedures. The collection of quality control samples is essential in order to provide confidence in the results of a sampling program, and is part of the overall quality assurance program.

4.2. Associated documents

Sampling design and preparation: sampling scope and design.

Physical and chemical assessment:

- Manual collection of surface water samples (including field filtration)
- Background to water quality sampling using automated sampling equipment
- Water quality sampling using automated sampling equipment
- Chlorophyll a sample collection methods
- Collection and preservation of sediment
- Guidance on the sampling of groundwaters

Biological assessment: Direct toxicity assessments

4.3. Introduction

The number and type of quality control samples collected will depend on the type of analysis being undertaken, the number of samples being collected and the level of contamination expected. For example, blank samples may be more important when assessing low levels of environmental contamination (e.g. ultra-trace metals in water) than when assessing industrial waste.

Field quality control samples can be used to assess and estimate:

- Whether a contaminant has been introduced into a sample during the sample collection, sample transport or in the laboratory analysis.
- The accuracy of the result. Accuracy refers to how close a result is to its true value.
- The precision within the results of a set of analyses. Precision relates to the repeatability of a result—the closeness of the results of multiple analyses to each other. It gives no indication of a results relationship to a true value.

The number of quality control samples should be defined during the sampling design process, and acceptance criteria should be determined during this phase. Acceptance criteria are the agreed upon limits that ensure data are of acceptable quality. If the quality control results are outside the acceptance criteria, investigations should be undertaken, and in some cases data will have to be rejected.

Note: The analytical method used for the quality control samples should be exactly the same as used to analyse other samples from the sampling program.

4.4. Quality control samples

4.4.1. Blanks

Blanks are used to trace sources of contamination that may be introduced into a sample from the sampling process, sample transportation, or from laboratory sources of contamination. The number of blanks collected for each sampling program will depend upon the programs objectives and size, and the types and concentrations of analytes being measured. Blanks should be analysed for the same analytes as all other samples. Blank water should be ultra-pure MilliQ water provided by the analytical laboratory, or if from another source routinely tested to show no contamination is present. Guidance on the number of blank samples is provided in Table 1. Some types of blanks are outlined below:

- **Container blank:** used to quantify and trace contamination problems associated with the sample containers and preservation. A bottle is filled with ultra-pure MilliQ supplied by the analytical laboratory, preservative is added (if required), and the sample is stored for the same time as the samples for analysis.

- **Trip or transport blanks:** used to estimate contamination of a sample from shipping and laboratory sources of contamination. A bottle is filled with ultra-pure MilliQ, preservative is added (if required), and the sample is transported using the same procedures as the samples. These blanks are generally used for assessing volatile contamination; however, it may also be appropriate to include field blanks for all analytes if using ice for shipping, and there is a chance of melted water coming into contact with sample bottles and very low levels of contaminants are being measured.
- **Field blanks:** used to estimate contamination of a sample during the collection procedure. Field blanks are prepared in the field in the same manner as the sample. A bottle is filled in the field with ultra-pure MilliQ water, preservative is added (if required), and for example, if a sample is field filtered, the field blank will also be filtered. It is recommended that water is supplied by the testing laboratory, or if produced in house, undergoes testing to ensure it is contaminant free.
- **Rinsate/Equipment blank:** used where an analyte free liquid is poured over or through decontaminated field sampling equipment to assess potential contamination from the equipment. Generally, ultra-pure MilliQ water is used, but other liquids may be more appropriate (e.g. analytical grade hexane if testing for hydrocarbons or organics).

Note: Distilled water purchased from retailers is not adequate for use as a blank.

4.4.2. Other field quality control samples

- **Trip spikes.** Particularly useful for volatile compounds. Prior to field trip, clean analyte free water spiked with a known concentration of compound of interest and taken to field and returned unopened for analysis.
- **Duplicate or triplicates:** duplicate or triplicate samples are obtained by splitting a sample into two or by taking subsamples from the collection container. They may also be collected in the field by sampling at exactly the same time and place. Duplicate results provide an estimate of the error associated with the subsampling/splitting process and laboratory analysis, and are a measure of precision. Duplicate samples should be sent in as blind samples to the laboratory. Depending upon the individual circumstances of the sampling program, it is recommended that a subset of duplicate or triplicate samples be sent to a second laboratory to confirm the primary laboratory's results.
- **Certified reference material (CRM):** certified reference materials are available for some analytes in some matrixes. Certified reference material can be used to assess laboratory accuracy and precision. Samples should be sent in as a blind sample. The concentration of analytes in the CRM should be in the range of the analytes expected in the environmental samples.

Note:

A blind sample is a sample named so that it is indistinguishable from the other samples. This means the source and chemical composition of the samples are not known to the analyst. Blanks, duplicates and CRM are often used as blind samples. Blind samples can determine variability within a laboratory or bias and variability between two or more laboratories.

Replicates: replicate samples are obtained by collecting two or more samples across a site at the same time. Replicate samples provide estimates of the sample variability, including experimental sampling error and analytical error. The number of replicate samples collected depends upon the program objectives. See Sampling design and preparation—sampling scope and design for further information.

Table 1: Guidance on the frequency of collection and purpose of quality control samples.

Quality Control Sample	Number of quality control samples to be collected	Notes
Container Blank	One per trip.	
Field Blank	One per field team per trip or one per 20 samples.	If sampling in a particularly dirty environment and for trace or ultra-trace concentrations of analytes, it may be useful to collect more field blanks (e.g. one per team per day).
Trip/Transport Blank	For volatile organic contaminants (VOC) - one per cooler box.	

Rinsate/Equipment Blank	One per field team per trip.	
Duplicates (triplicates)	For waters: one per 10 samples for primary laboratory, one per twenty samples to the second laboratory (or one per program when there are less than 20 samples). For soils: one per 20 samples.	Assesses precision of the results within a laboratory and between laboratories.
Trip spike	For VOC – one per trip.	Tests the loss volatile compounds.
Certified Reference Material	Ad hoc. One per large sampling project.	Evaluates laboratory accuracy and precision. Should be sent in as a blind sample.

4.5. Guidance on assessing quality control data

Quality control data should be assessed immediately upon receipt of data from the laboratory to ensure any problems indicated from the quality control program can be investigated as soon as possible.

4.5.1. Blanks

Any reported concentrations of inorganic or organic contaminants in blanks should be investigated immediately. Collection of adequate blank data should indicate if the source of contamination is the container, equipment, field, transport or the laboratory.

4.5.2. Duplicates

The assessment of duplicates is commonly undertaken by expressing the duplicate results as the Relative Percent Difference (RPD). As a rule of thumb, a RPD of $\leq 20\%$ may indicate an acceptable result for duplicate aqueous samples (Equation 1), provided the result is five to ten times the limit of reporting (LOR). In those circumstances where the result is close to the LOR, RPD may exceed 20%. However, the acceptable RPD can be strongly influenced by the analyte and matrix. Therefore, specific acceptance criteria should be discussed with the laboratory prior to the sampling program commencing. The Australian Standard for Soils (AS 4482.1-2005), which can be applied to sediments, gives an acceptance criteria RPD of 30-50%, noting that the variation is higher for organic than inorganic analyses.

$$RPD = \frac{|C_1 - C_2|}{\left(\frac{C_1 + C_2}{2}\right)} \times 100$$

Equation 1

Where: RPD is relative percentage difference

C1 is the concentration of analyte from sample 1

C2 is the concentration of analyte from sample 2.

4.5.3. Certified reference material

To assess how far away a result is from a true result using field spikes and certified reference material (CRM), the percent recovery is typically used (Equation 2).

$$\%R = \frac{(X)}{(K)} \times 100$$

Equation 2

Where: %R is recovery as %

X is the measured concentration

K is the known or accepted/true concentration.

For CRM, acceptable recovery ranges are provided by the supplier. Acceptable % recovery ranges for field spikes are dependent upon the analyte and matrix, and should be discussed with the laboratory.

4.5.4. Total versus dissolved

A further quality control check can be undertaken by comparing the total concentrations with the dissolved² concentrations where applicable. This is most appropriate where the sample used to measure the dissolved concentration has been subsampled from the one collected to measure total concentration. If samples are collected separately, there may be variability in environmental conditions at the exact time of collection.

In general, the dissolved concentration should be less than the total concentration. However, as there is a level of uncertainty associated with any result, the dissolved concentration may exceed the total concentration. An example is given below in Table 2. The uncertainty of the method can be obtained from the testing laboratory.

Table 2: Worked examples of total versus dissolved metal check for zinc (based on an example method uncertainty of 20%). This is only valid if the dissolved metals were subsampled from the same sample as the total metals.

Results	Notes	Recommended action
Total zinc = 1.4mg/L (1.1–1.7mg/L) (20% uncertainty is ± 0.3mg/L) Dissolved zinc = 0.7mg/L (0.6-0.8mg/L) (20% uncertainty is ± 0.1mg/L)	Total zinc is greater than dissolved zinc	No investigation required
Total zinc = 1.4mg/L (1.1–1.7mg/L) (20% uncertainty is ± 0.3mg/L) Dissolved zinc = 1.6 mg/L (1.3-1.9mg/L) (20% uncertainty is ± 0.3mg/L)	Total zinc is less than dissolved zinc, but the difference is within the uncertainty of the method.	No investigation required
Total zinc = 1.4mg/L (1.1–1.7mg/L) (20% uncertainty is ± 0.3mg/L) Dissolved zinc = 2.2mg/L (1.8-2.6mg/L) (20% uncertainty is ± 0.4mg/L)	Total zinc is less than dissolved zinc and the difference is greater than the method uncertainty.	Investigation required

4.6. References and additional reading

AS 4482.1-2005: Guide to the investigation and sampling of sites with potentially contaminated soil – Non-volatile and semi-volatile compounds.

AS/NZS 5667.1:1998 (R2016). Water quality – Sampling – Guidance on the design of sampling programs, sampling techniques and the preservation and handling of samples.

National Association of Testing Authorities (NATA) 2004, Technical Note 17 — October 2013, Guidelines for the validation and verification of quantitative and qualitative test methods. Available from:

http://www.nata.com.au/nata/phocadownload/publications/Guidance_information/tech-notes-information-papers/technical_note_17.pdf

United States Geological Survey (USGS) 2006, National Field Manual for the Collection of Water-Quality Data, Chapter 44, Collection of water samples. Available from:

https://water.usgs.gov/owq/FieldManual/chapter4/pdf/Chap4_v2.pdf

² Dissolved samples are operationally defined as samples that have been filtered through a 0.45µm filter.

Permits and approvals

Version: February 2018

5. Permits and approvals

5.1. Purpose and scope

This document provides information on the consideration of permit and/or approval requirements. Prior to commencing any sampling activities, all necessary permits and approvals must be obtained. Permits and approvals may be required to conduct activities involving animals or protected areas (for example National Park/Regional Park, State Forest, State Marine Park or declared fish habitat area). Failure to do so may be a breach under the Nature Conservation Act 1992, Animal Care and Protection Act 2001, Fisheries Act 1994, Recreation Areas Management Act 2006 or the Sustainable Planning Act 2009.

5.2. Required permits and approvals

More than one permit and/or approval may be required for any given activity. For more information on permits and approvals that may be required, contact the following areas:

- Protected wildlife and protected areas (including activities within Queensland's national parks and forests): Queensland Government, <http://www.qld.gov.au/environment/parks/permits>
- Fisheries permits: Department of Agriculture and Fisheries, <https://www.business.qld.gov.au/industry/fisheries/commercial-fishing/licences-and-fees/commercial-fishing-licences/fishing-permits>
- Using animals for scientific purposes: Department of Agriculture and Fisheries, <https://www.daf.qld.gov.au/animal-industries/welfare-and-ethics/using-animals-for-scientific-purposes>
- Education, research or monitoring in a declared fish habitat area or involving the removal, destruction or damage of marine plants: Department of Agriculture and Fisheries, <https://www.daf.qld.gov.au/fisheries/habitats/fisheries-development/self-assessable-codes>
- If your sampling and/or research is to occur within the Great Barrier Reef Marine Park you will likely require a permission from the Great Barrier Reef Marine Park Authority. Enquiries about Marine Park permits should be sent to assessments@gbbrmpa.gov.au.

5.3. Granted permits or approvals

Once a permit or approval has been granted, it is often necessary to contact the authorising department to notify the district staff of the date and time of your visit. This is done prior to entering the protected area and/or commencing the approved activity. For further information regarding notification requirements, check the permit or contact the relevant authority. There may be additional requirements associated with permit approvals, such as annual reporting to be provided to the relevant department regarding activities under the permit.

5.4. Permission to access land

In addition to permits and approvals gained under legislation, it is vital permission is gained prior to accessing any privately owned land for undertaking sampling. You may also need to consider additional biosecurity requirements such as vehicle/machinery/equipment hygiene inspection reports (used for preventing the spreading of weeds on properties). See Department of Agriculture and Fisheries for more information (<https://www.daf.qld.gov.au/plants/weeds-pest-animals-ants/weeds/preventing-weed-spread/cleanup>)

Control and reference sites

Version: February 2018

6. Control and reference sites

6.1. Purpose and scope

'Control sites' and 'reference sites' are terms that are frequently used to describe sites that can be used to assess the impacts of a disturbance or pollution events. However, there is often considerable confusion between the two terms. This document describes the terms as used in a regulatory context under the Environmental Protection Act 1994 and the Environmental Protection (Water) Policy 2009 (EPP Water).

6.2. Introduction

When assessing potential environmental impacts it is necessary to measure one or more indicators that will provide information (generally numerical quantities or qualitative ranks) about the environmental condition at the potentially impacted site, or test site, and compare these measurements against similar measurements collected in the absence of impact or disturbance.

Control sites can be described as 'monitoring sites that are identical in all respects to the site being assessed (sometimes called the test site) except for the disturbance' (Section 3.1.4.1; ANZECC & ARMCANZ 2000). Control sites are usually upstream, off-stream or in another location in the vicinity of the proposed activity or wastewater release, and therefore, not impacted by the activity or wastewater release. Values for one or more indicators that may be considered controls could also include those data collected prior to an activity commencing, provided that an adequate quantity of data is available. The use of control sites is favoured for compliance and regulation assessment.

Reference sites, in contrast, are those that are considered to represent pristine environments. In practice, there are very few truly pristine environments in Queensland, so minimally disturbed and best available sites are often used as proxies for pristine condition. All attempts should be made to identify pristine sites before using minimally disturbed and best available sites as reference sites. Under the EPP Water, reference site data are used as the basis for the development of the Queensland Water Quality Guidelines (QWQG) (DEHP 2009) and Scheduled Water Quality Objectives³. Water Quality Objectives, in particular, are intended as a benchmark for improvement of water quality on a catchment scale, with the aim of returning a system to a more 'natural' condition.

6.3. Consideration in selecting control and reference sites

It is important to note that as control or reference sites are used for different purposes, a site that may be suitable as a control or reference site for one indicator may not be relevant for another. For example, a pristine site may be suitable for assessing the natural condition of macroinvertebrates or water quality in an area, but the existence of a major in-stream barrier downstream that prevents migration of fish, such as a dam, may mean that the site is not suitable as a fish reference site.

As flow conditions impact significantly on water quality, control and reference data need to be collected at an appropriate site and relate to the flow regime at the time of collection. Further, consideration must be given to the number of control or reference sites needed for adequate assessment (i.e. by using power analysis where possible).

6.4. Reference site criteria used for bioassessments

Protocols outlined in the River Bioassessment Manual (Davies 1994) describe the process for selecting reference and test sites. From this, the Queensland Department of Natural Resources and Mines (DNRM 2002) composed a set of ten selection criteria that should be used to determine whether or not a site is in reference condition for biological assessments. Each criterion relates to an anthropogenic activity that has the potential to modify the

³ Water Quality Objectives are *scheduled* pursuant to the provisions of the Environmental Protection (Water) Policy 2009, which is subordinate legislation under the Environmental Protection Act 1994. They are provided in Schedule 1 documents that list the Environmental Values and Water Quality Objectives for waters in Queensland (<https://www.ehp.qld.gov.au/water/policy/>).

natural condition of the freshwater ecosystem. Where pristine sites are not available, minimally disturbed and best available sites must be used. By generating a numerical categorisation of sites, this approach ensures that reference sites are comparable across programs.

The criteria presented here have been modified slightly from DNRM (2002), resulting in the following 11 criteria:

1. Agriculture and forestry
2. Grazing intensity
3. Sand/gravel extraction
4. Upstream urban areas
5. Point source pollution
6. Barriers – impact on biota
7. Flow regime alteration
8. Riparian and valley flat vegetation
9. Weed species in riparian zone
10. Bankside erosion / deposition
11. Instream habitat alteration

These criteria are given a score between 1 and 5 representing the following categories:

1. Extreme impact
2. Major impact
3. Moderate impact
4. Minor impact
5. No impact

The ideal 'reference condition' site would score 5 in all 11 criteria (i.e. no impact). For a site to be classified as being of 'reference condition' it must score a 4 or 5 in each of the 11 criteria (i.e. minimally disturbed and/or best available site). If the impacts are unknown, the assessors must seek further information before scoring. Where a site receives a score of less than 5, comments must be provided to justify the score. As much information as possible should be provided. The reference site criteria used to develop guidelines for physico-chemical water quality indicators in the QWQG (DES 2009) use a modified version of the eleven reference site criteria and are discussed further in Section 5.

Diverse sources of information, including previous knowledge of the catchment and maps, can be used to determine a score for some of the criteria and select potential reference sites. If possible, before a sampling program commences, it is recommended that reconnaissance surveys be used to confirm the suitability of potential reference sites. Potential sites may be chosen by inspecting a large area prior to completing the reference criteria. Alternatively, assessment against the reference condition selection criteria can be completed upon arrival at a site prior to sampling - the assessors would need to conduct an inspection of the site (i.e. walk several hundred metres along the stream reach).

It is also recommended that the scores be reviewed again after sampling/surveying is completed, because the team members will have a better understanding of the site in question. This is particularly important when conducting riparian surveys where a much larger area is covered as part of the survey compared to the initial site inspection. The knowledge obtained from the survey may lead to an alteration of the scores.

Although the criteria are soundly based, in reality it may be difficult to find any sites that adequately meet these criteria. Because of this, flexibility may be required when applying the criteria in some situations. Seek expert advice to determine the extent (if any) to which the criteria may be relaxed in situations where ideal reference sites are lacking.

Scoring against the criteria is somewhat subjective. The variability in individual scores can be minimised if training is conducted by experienced staff and through the use of the example field sheets with descriptions of impact levels for each criterion. An example field sheet is presented in Appendix 1. To assist scoring, the field sheet includes examples of each of the five levels of impact for each criterion. Appendix 2 provides examples of possible impacts for each of the selection criteria. More than one person must complete this form, and all assessors should be in agreement with the final scores.

A data-driven method for selecting reference sites for stream bio-assessments of freshwater fish are described by Rose et al. (2016) and may be used if suitable.

6.5. Reference site criteria used for physico-chemical water quality indicators

The QWQG (DES 2009) provide a different set of reference site criteria to those outlined above, and these are to be applied specifically for physico-chemical water quality indicators. The QWQG define a reference site as 'a site whose condition is considered to be a suitable baseline or benchmark for assessment and management of sites in similar water bodies'. The criteria adopted to choose reference sites for physico-chemical indicators in the QWQG

are shown in Table 1. The QWQG provide some flexibility in defining the reference condition. It states:

'The reference condition concept can also be applied to more disturbed systems. For example, in an urban situation it might be useful to use the least disturbed urban creek sites to derive reference values and guidelines to be applied to other urban creeks. This would provide a realistic expectation of quality in an urban situation whereas use of largely undisturbed reference sites for highly disturbed systems might create unachievable water quality expectations.'

Through existing state government monitoring programs, a number of minimally disturbed reference sites have already been identified throughout Queensland. These are listed in Appendix F of the QWQG. Care should be taken when applying sites previously deemed as minimally disturbed reference sites as their status can easily change over time. Further assessment of these sites should be made using the criteria listed above.

Table 3: Criteria for reference sites for physico-chemical indicators

Freshwaters	
1	No intensive agriculture within 20km upstream. Intensive agriculture is that which involves irrigation, widespread soil disturbance, use of agrochemicals and pine plantations. Dry-land grazing does not fall into this category.
2	No major extractive industry (current or historical) within 20km upstream. This includes mines, quarries and sand/gravel extraction.
3	No major urban area (>5000 population) within 20km upstream. If the urban area is small and the river large this criterion can be relaxed.
4	No significant point source wastewater discharge within 20km upstream. Exceptions can again be made for small discharges into large rivers.
5	No significant point source wastewater discharge within 20km upstream. Exceptions can again be made for small discharges into large rivers.
Estuaries	
1	No significant point source wastewater discharge within the estuary or within 20km upstream. Exceptions can again be made for small discharges into large rivers.
2	No major urban area (>5000 population) within 20km upstream. If the urban area is small and the river large, this criterion can be relaxed.

6.6. References and additional reading

Bailey, RC, Norris RH and Reynoldson, TB 2004, *Bioassessment of Freshwater Ecosystems: Using the Reference Condition Approach*. Springer Science + Business Media Inc. New York. USA.

Davies, PE 1994, *National River Processes and Management Program Monitoring River Health Initiative: River Bio-assessment Manual*, Commonwealth Environmental Protection Agency, Canberra.

DES 2009, *Queensland Water Quality Guidelines, Version 3*, ISBN 978-0-9806986-0-2, Department of Environment and Heritage Protection, Queensland Government, Brisbane, Available from <https://www.ehp.qld.gov.au/water/pdf/water-quality-guidelines.pdf>.

DNRM 2002, *Australia-Wide Assessment of River Health: Queensland AusRivAS Sampling and Processing Manual*, Monitoring River Health Initiative Technical Report Number 12, Environment Australia, Canberra.

Rose, PM, Kennard, MJ, Sheldon, F, Moffatt DB, and Butler GL 2016, 'A data-driven method for selecting candidate reference sites for stream bioassessment programs using generalised dissimilarity models', *Marine and Freshwater Research*, 67, 440-454.

Appendix 1 Reference condition selection criteria field sheet

Project Name: _____ Project Code: _____ Run Number: _____ Photos taken (circle one): YES/NO Date: _____

Assessors: _____ Site Number: _____ Site Name: _____

Possible Impacts	5	4	3	2	1	Score
	(No Impact)	(Minor Impact)	(Moderate Impact)	(Major Impact)	(Extreme Impact)	
1. Agriculture and forestry*	No impact	Present but level of impact is barely discernible	Evident; however, not severe and/or widespread	Obvious impact to stream; moderate and/or widespread	Severe and widespread; impact obvious	
2. Grazing intensity	No impact	Present but level of impact is barely discernible	Impacts evident; however, not severe and/or widespread	Obvious impact to stream; moderate and/or widespread	Severe and widespread; impact obvious	
3. Sand/gravel extraction*	No evidence or prior knowledge of extraction	Small scale historical extraction	No current extraction; large historical extraction	Current small scale/localised extraction	Current and widespread extraction	
4. Upstream urban areas*	No impacts from urbanisation	Possible impacts caused from urbanisation	Definite impacts caused from urbanisation	High impacts caused from urbanisation	Extreme impacts caused from urbanisation	
5. Point source pollution*	Nil point source pollution	Low volumes of point source pollution discharged	Low to moderate volumes of point source pollution discharged	Moderate to high volumes of point source pollution discharged	High to extreme volumes of point source pollution discharged	
6. Barriers-impact on biota*	No artificial barriers in basin which will affect the site	Few small upstream barriers; not within impoundment	Many small barriers; site not within impoundment	Multiple small barriers; large barriers upstream; within small impoundment	Large barriers upstream; within large impoundment	
7. Flow regime alteration*	Seasonal flow regime natural	Seasonal flow regime not obviously altered	Flow regime altered	Flow regime obviously altered	Flow regime highly modified	
8. Riparian and valley flat vegetation#	Streamside vegetation unaltered	Vegetation slightly modified	Obvious modification	Highly modified vegetation	Severe modification	
9. Weed species in riparian zone#	Weed species absent or insignificant	Few introduced species present; disturbance is minor	Some introduced species present; disturbance is moderate	High percentage of introduced species; disturbance is high	Vegetation dominated by introduced species; extreme disturbance	
10. Bankside erosion / deposition#	No evidence of erosion beyond natural	Slightly more than natural levels of erosion	Moderate levels of unnatural erosion	High levels of erosion	Extreme erosion	
11. Instream habitat alteration#	Instream habitats of natural appearance and diversity	Barely discernible impacts	Moderate modifications to instream habitats	Highly modified modifications to instream habitats	Severe modification of instream habitats	
Total						
Is site in reference condition i.e. all scores ≥ 4? (Yes or No)						

Notes:

* the level of impact will generally decrease as the distance from the source of impact increases

some of these variables will vary between and within catchments - compare with that which should be expected (i.e. natural).

If the impacts are unknown, seek further information before scoring.

More than one person must complete this form.

Provide comments for all criteria scoring <5 on the following page.

Selection criteria comments

Selection Criteria	Comments
--------------------	----------

SC1

SC2

SC3

SC4

SC5

SC6

SC7

SC8

SC9

SC10

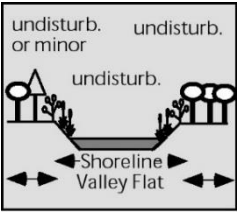
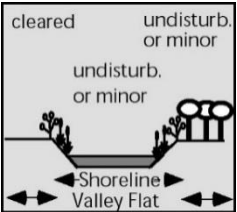
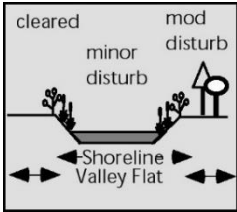
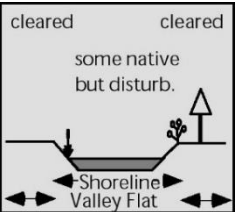
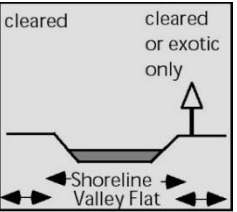
SC11

When filling in comments, provide as much information as possible, such as:

- identify desk based resources used in assessment
- details of type and source of impacts including approximate distances from those sources
- specific comments relating to assessments for each indicator, specifically when giving a score less than 5.

Appendix 2 Example of possible impacts for each selection criteria

Possible impacts and examples	5 (No impact)	4 (Minor impact)	3 (Moderate impact)	2 (Major impact)	1 (Extreme impact)
1. Agriculture and forestry	<ul style="list-style-type: none"> No agriculture and/or forestry 	<ul style="list-style-type: none"> Dryland cropping and native species plantation with substantial vegetation buffer zones >30m present 	<ul style="list-style-type: none"> Moderate dryland cropping and native species plantation with narrow/fragmented vegetation buffer zones <30m Pine plantation and irrigated cropping with substantial vegetation buffer zone >30m 	<ul style="list-style-type: none"> Pine plantation and irrigated cropping with narrow/ fragmented vegetation buffer zones <30m Dryland cropping and native species plantation with no vegetation buffer-zones present 	<ul style="list-style-type: none"> Widespread soil disturbance extending to top of stream banks Extensive use of agrochemicals
2. Grazing intensity	<ul style="list-style-type: none"> No grazing 	<ul style="list-style-type: none"> Light grazing in natural forest with limited and/or infrequent stock access to stream. Moderate grazing pasture. 	<ul style="list-style-type: none"> Moderate grazing in natural forest with widespread and/or frequent stock access to river Moderate grazing pasture, with narrow or fragmented veg. buffer zones <30m Heavy grazing, dairy, with substantial vegetation buffer-zone >30m 	<ul style="list-style-type: none"> Heavy grazing or dairy with narrow/ fragmented vegetation buffer zones <30m Moderate grazing pasture with no vegetation buffer-zones present 	<ul style="list-style-type: none"> Heavy grazing, dairy, pine plantation and irrigated cropping with no vegetation buffer zones present
3. Sand/gravel extraction	<ul style="list-style-type: none"> No knowledge of upstream and/or downstream extraction Note: impacts must be present at site, rather than impacts that could be possibly occurring. 	<ul style="list-style-type: none"> Small scale historical extraction with impacts barely apparent Current small scale floodplain extraction 	<ul style="list-style-type: none"> Historical instream extraction, with impacts still apparent Current large scale floodplain extraction 	<ul style="list-style-type: none"> Current small scale instream extraction 	<ul style="list-style-type: none"> Current widespread extraction
4. Upstream urban areas	<ul style="list-style-type: none"> No urbanisation upstream 	<ul style="list-style-type: none"> Small town on large stream; few upstream towns 	<ul style="list-style-type: none"> Medium town (pop 3000 to 10,000) on small stream (width<30m), >10km upstream; few upstream towns Small town on large stream, <10km upstream; many upstream towns 	<ul style="list-style-type: none"> Medium town on small stream, <10km downstream; many upstream towns Stream >10km from large town (pop>10,000) 	<ul style="list-style-type: none"> Stream <10km from large town
5. Point source pollution	<ul style="list-style-type: none"> Nil point source pollution upstream Note: score will vary significantly depending upon the type of pollutant discharged. Examples include sewage, road drainage, industrial waste, thermal pollution, etc. When applicable, write down the type of pollutant discharged. 	<ul style="list-style-type: none"> Low volumes of effluent into large (width >30m) permanently flowing stream 	<ul style="list-style-type: none"> Low volumes of effluent into small (width <30m) permanently flowing streams Moderate volumes of effluent into large permanently flowing stream 	<ul style="list-style-type: none"> High volumes of effluent into large permanently flowing stream Moderate volumes of effluent into small permanently flowing stream Low volumes of effluent into temporary stream during flowing periods 	<ul style="list-style-type: none"> High volumes of effluent into permanently flowing stream Discharge into temporary stream during no/low flow

Possible impacts and examples	5 (No impact)	4 (Minor impact)	3 (Moderate impact)	2 (Major impact)	1 (Extreme impact)
6. Barriers-impact on biota	<ul style="list-style-type: none"> No barriers upstream or within 10km downstream from site Note: barriers can be artificial such as dams/weirs but should also include road crossings and other obstructions to the passage of biota. 	<ul style="list-style-type: none"> Few small (<2m high) barriers upstream from site Site not within impoundment No barriers within 10km downstream from site 	<ul style="list-style-type: none"> Many small barriers upstream from site Flow regime obviously altered Site not within impoundment 	<ul style="list-style-type: none"> Large dam/weir or artificial barriers >10km upstream Multiple upstream small dams/weirs or artificial barriers Instream habitat obviously altered (e.g. artificial riffles created, dried out or drowned) Within small impoundment with stable water levels 	<ul style="list-style-type: none"> Large dam/weir <10km upstream from site Within impoundment with no flow or highly fluctuating water levels
7. Flow regime alteration	<ul style="list-style-type: none"> No flow alteration due to abstraction, impoundments or water releases Or "No knowledge" of any such effects 	<ul style="list-style-type: none"> Some abstraction from large stream Base flow stopped or decreased 	<ul style="list-style-type: none"> Occasional releases supplement base flow Abstraction high relative to stream size decreasing or stopping flow Low abstraction during low/no flow periods Base flow stopped 	<ul style="list-style-type: none"> Frequent releases supplement base flows Large abstraction from a permanent small stream obviously reducing/stopping flow and water level Moderate abstraction during low/no flow periods Base or medium flow stopped or reduced 	<ul style="list-style-type: none"> Seasonality of flow regime reversed by dams/weirs stopping flood flows and frequently/continuously Releasing supplemental base flows, site severely affected by abstraction or regulation Large abstraction during low/no flow periods
8. Riparian and valley flat vegetation	<p>Valley flat vegetation—native vegetation present on BOTH sides of the river with a virtually intact canopy. Shoreline vegetation—native vegetation on BOTH sides of the river is generally in good condition. Any disturbance is minor.</p> 	<p>Valley flat vegetation—agricultural land and/or cleared on ONE side; native vegetation on the other in reasonably undisturbed state. Shoreline vegetation—native vegetation on BOTH sides with canopy intact or with native species widespread and common in the shoreline zone.</p> 	<p>Valley flat vegetation—agricultural land and/or cleared on ONE side; native vegetation on the other clearly disturbed. Shoreline vegetation—bank vegetation moderately disturbed though native species remain.</p> 	<p>Valley flat vegetation—agriculture and/or cleared land BOTH sides. Shoreline vegetation—native vegetation present, but it is modified on BOTH sides.</p> 	<p>Valley flat vegetation—agriculture and/or cleared land BOTH sides. Shoreline vegetation—absent or severely reduced. Vegetation present is extremely disturbed.</p> 
9. Weed species in riparian zone	<ul style="list-style-type: none"> Weed species absent (0%) 	<ul style="list-style-type: none"> Weed species <20% Disturbances from presence of weeds is minor 	<ul style="list-style-type: none"> Obvious presence of exotic species (20-40%) 	<ul style="list-style-type: none"> High percentage of exotic species in riparian zone (40-60%) 	<ul style="list-style-type: none"> Riparian zone dominated by exotic vegetation (>60%)
10. Bankside erosion and deposition	<ul style="list-style-type: none"> Riparian zone and stream banks in natural condition No unnatural erosion 	<ul style="list-style-type: none"> Riparian zone and stream banks with barely discernible erosion impacts Infrequent, small areas (<20%) of unnatural erosion 	<ul style="list-style-type: none"> Riparian zone and stream banks with erosion impacts Moderate sized areas (20-40%) of unnatural erosion 	<ul style="list-style-type: none"> Riparian zone and stream banks with obvious erosion impacts Extensive areas (40-60%) of unnatural erosion 	<ul style="list-style-type: none"> Riparian zone and stream banks with severe erosion impacts Majority (>60%) of area unnaturally eroded
11. Instream habitat alteration	<ul style="list-style-type: none"> Diverse number of naturally occurring instream habitats in natural condition (e.g. some macrophyte growth, little algal growth, abundant coarse woody debris) No evidence of stream bed aggregation or degradation 	<ul style="list-style-type: none"> Partial loss of some habitats and alteration to condition (e.g. increased macrophyte growth, algal growth, some loss of woody debris) If present then only slight degradation or aggregation Exhibits few of the degraded 	<ul style="list-style-type: none"> Limited loss of some instream habitats (from drying, drowning, silting, scouring etc.) and alteration to conditions Moderate algal and/or macrophyte growth may extensively cover some areas of reach 	<ul style="list-style-type: none"> Widespread loss of instream habitats (from drying, drowning, silting, scouring etc.) and alteration to conditions Extensive algal and/or macrophyte growth smothering areas of reach Coarse woody debris removed 	<ul style="list-style-type: none"> Dominated by only one habitat (due to drying, drowning, filling or scouring), conditions highly modified Extensive macrophyte and algal growth chokes whole reach No woody debris Substrate smothered with deep

Possible impacts and examples	5 (No impact)	4 (Minor impact)	3 (Moderate impact)	2 (Major impact)	1 (Extreme impact)
	<ul style="list-style-type: none"> Note: Degraded symptoms include: bed shallowing or deepening; bed erosion; steepening/undercutting banks; exposure of bridge bases; headcut or nickpoint; steep/mobile riffles. 	symptoms	<ul style="list-style-type: none"> Some coarse woody debris removal Exhibits more than a few symptoms Moderate degradation 	<ul style="list-style-type: none"> Exhibits more than a few symptoms Moderate to severe degradation 	<ul style="list-style-type: none"> layer of rotting vegetation (such as in Para grass choked streams) Exhibits many of the degradation symptoms listed. Severe degradation

Background information on the Global Positioning System and handheld units

Version: February 2018

7. Background information on the Global Positioning System and handheld units

7.1. Purpose and scope

This document will help develop a basic understanding of handheld Global Positioning System (GPS) units and the methods they use to calculate a position. It also covers factors that may affect the accuracy of position calculations and methods to mitigate the effect of those factors.

7.2. Associated documents

Sampling design and preparation: Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection

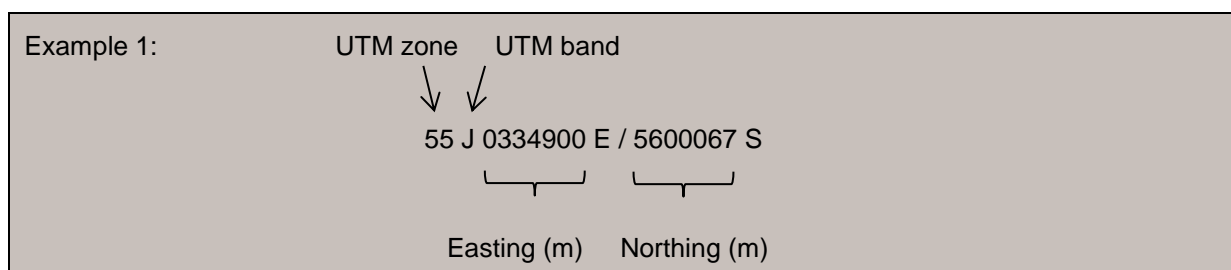
7.3. Introduction

Global Positioning System (GPS) receiver units are important tools for determining accurate locations for field sampling sites.

There are two main types of coordinate systems—spherical coordinates expressed as latitudes and longitudes (referred to as geographic coordinates) and projected coordinates (usually expressed in metres relative to the origin point of the projected plane). Geographic coordinate systems establish a position based on the earth's approximately spherical surface, whereas projected coordinate systems are a way of presenting the three dimensional (3D) data of the earth's surface on a two dimensional (2D) plane.

The World Geodetic System 1984 (WGS84) is a commonly used datum in GPS units world-wide. A large number of 'localised' datums also exist, for example the Geocentric Datum of Australia 1994 (GDA94). GPS units can convert the WGS84 position to other datums. Datums are typically expressed in decimal degrees or degrees minutes and seconds of arc.

The Universal Transverse Mercator (UTM) projection is one method used world-wide to project the earth's surface onto maps. A key feature of the UTM projection is that it creates sixty zones around the earth, each six degrees in width (**Figure 4**). An example of how UTM coordinates are written using eastings and northings, as well as what each section relates to is shown below (Example 1). The current UTM projection is based on the WGS84 datum. Older maps and digital spatial data (including GPS coordinates) may be based on other datum, resulting in some discrepancies with position.



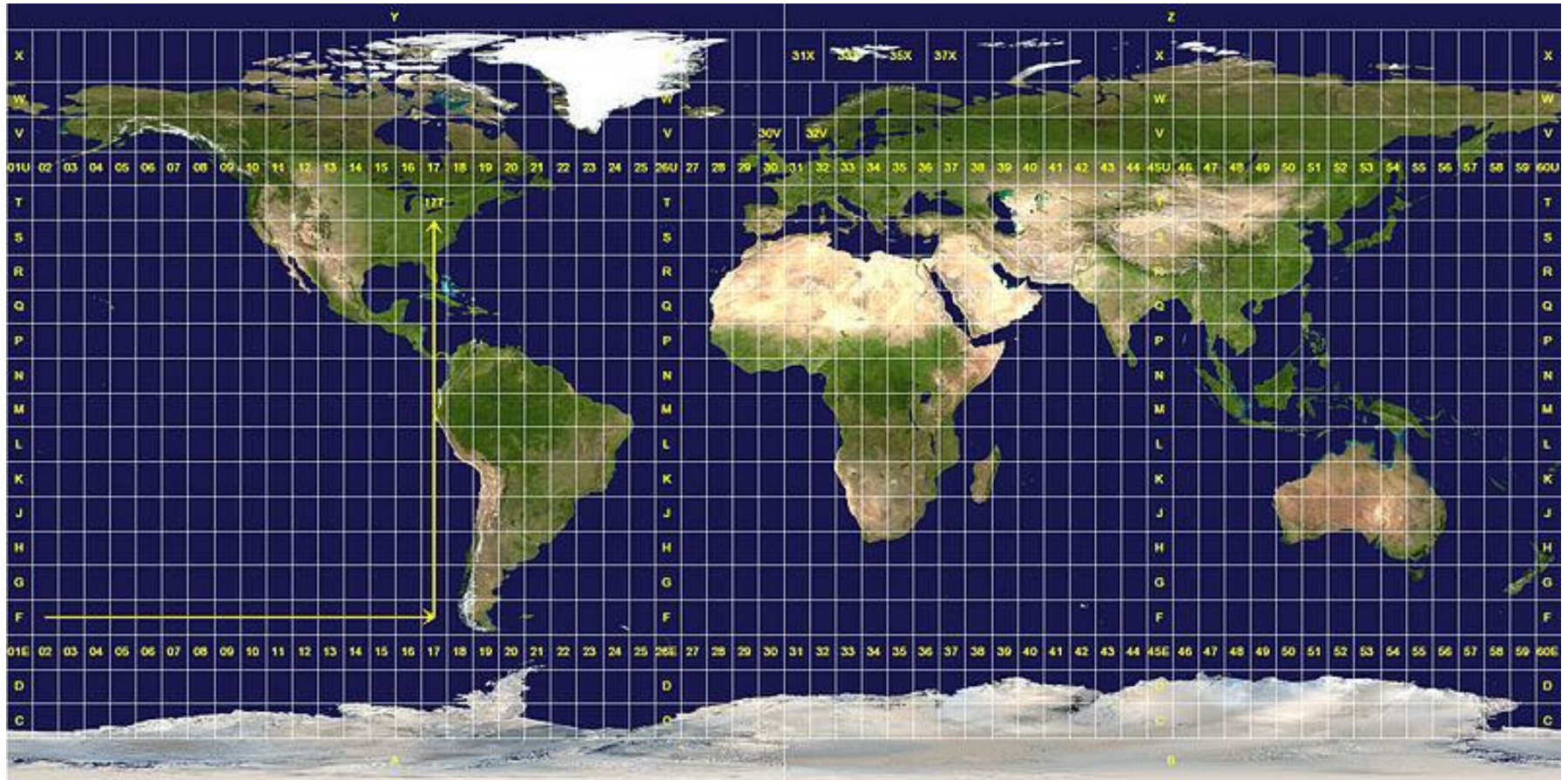


Figure 4: Universal Transverse Mercator (UTM) zones

"UTM-zones". Public Domain, Licensed under the Creative Commons Attribution-Share Alike 3.0 Unported licence. Wikimedia Commons – <https://commons.wikimedia.org/wiki/File:Utm-zones.jpg#/media/File:Utm-zones.jpg>.

7.4. How GPS receivers calculate positions

GPS satellites transmit a signal that includes the unique identifier for the satellite, its location in space and the time of the signal. This transmission is received by the GPS unit and used, in combination with signals from other satellites, to calculate the position of the GPS unit.

At least four satellites must be visible (above the horizon) to calculate a '3D position' (longitude, latitude, elevation). Generally, the greater the number of satellites used to calculate the position, the more accurate the calculated position is.

Most GPS units will indicate when it is able to calculate a 3D position or if it is still acquiring the satellites and either cannot provide a position or only provide a 2D position (latitude, longitude). It is recommended that you do not collect 2D positions because of the problem encountered with the altitude measurement. When you set the receiver to calculate 2D positions, you are replacing one satellite measurement (altitude) with a fixed measurement. If this altitude is incorrect, the latitude and longitude will also be incorrect. For example, if the fixed altitude is incorrect by 10 meters, the calculated horizontal measurements can be incorrect by 50 meters or more (Trimble Navigation Limited 2002).

The relative geometry of the satellites and the GPS unit plays an important part in the accuracy of the position solution. This effect is called Geometric Dilution of Precision (GDOP). GDOP refers to where the satellites are in relation to one another, and is a measure of the quality of the satellite configuration. It can magnify or lessen other GPS errors. In general, the wider the angle between satellites, the better the measurement.

7.5. GPS precision, accuracy and reliability

A GPS unit can produce precise, accurate and reliable positional data. For the purpose of this document, the term accuracy is used to describe all three factors outlined above.

The accuracy of the GPS unit's position calculation can be affected by a number of factors. Many of these factors can be managed by the GPS unit itself or through the users understanding of the operation of the GPS unit and their surroundings.

Most units display an 'accuracy' reading which partly indicates the degree of precision of the position calculation (**Figure 5**), but may not account for all sources of interference with the signals and so should be treated with caution. Generally, a basic handheld GPS unit will be relatively accurate and most modern units can calculate a 'horizontal' position to within 10m and a 'vertical' position (elevation) to within 50–100m.

Interferences to be aware of are:

- Vegetation canopy cover or nearby landscape features will affect the number of satellites being received by the GPS unit. The most accurate readings will generally be received when a clear view of the sky is available and signals are not at risk of interference.
- If the GPS unit is being used whilst stationary, it can (subject to other factors) provide a more accurate position calculation than if moving. Using the unit whilst completely stationary allows an improvement in the calculation through the unit being able to average its calculations over time.
- Holding a GPS unit close to the operator's body or within a vehicle is likely to affect 'line of sight' with satellites and affect the accuracy of the GPS reading.
- When a satellite is 'low' in the horizon, the signal must travel a longer distance through the ionosphere and troposphere. This could result in a weaker signal and greater interference. Most modern GPS units will only detect satellites that are greater than 15 degrees above the horizon to minimise these effects.
- The quality and position of the GPS unit's antennae can also be a factor. Many units can be fitted with an external (vehicle or high mounted) antennae to improve the ability of the unit to receive signals without interference.
- The signal-to-noise ratio (SNR) of a satellite is a measure of the signal strength or the amount of information content relative to the signal's noise. Noise refers to the degree of interference to the signal and can typically be caused by a weakening of the signal due to:
 - The signal having penetrated a barrier (e.g. vegetation cover)
 - Reflection of the signal off another surface, called multipath, such as buildings or even the operators body. Normally, accuracy errors from this source is less than 3m
 - The position of the satellite low in the horizon
 - Antennae quality.
- Unintentional radio frequency interference can be caused by TV transmitter signals, radar signals, and CB radio transmissions. Intentional interference is also possible. Radio frequency interference can cause complete loss of tracking, distorted signals, and incomplete data.

Table 1 outlines potential sources of GPS error.

Note: Ensure the GPS unit you use, and its ability to accurately calculate a position, matches the data quality or accuracy standard you require for the activity or task you are completing. The need for highly accurate positional data (e.g. to determine the exact cadastral boundaries of a property) will require specific equipment and appropriately qualified operators of that equipment.

Table 1: GPS error budget (taken from <http://www.montana.edu/gps/understd.html>)

Error source	Potential error	Typical error
Ionosphere	5.0 meters	0.4 meters
Troposphere	0.5 meters	0.2 meters
Ephemeris data	2.5 meters	0 meters
Satellite clock drift	1.5 meters	0 meters
Multipath	0.6 meters	0.6 meters
Measurement noise	0.3 meters	0.3 meters
Total	~ 15 meters	~ 10 meters

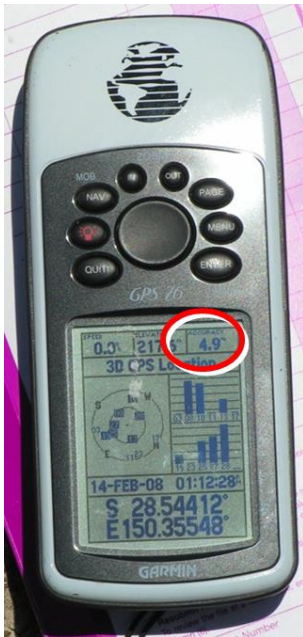


Figure 5: Most GPS units display an ‘accuracy’ reading

7.6. References and additional reading

Garmin Corporation 2000, [GPS Guide for Beginners](#), viewed 23 June 2002.

Trimble Navigation Limited 2010, [Trimble GPS Tutorial](#), viewed 23 June 2002.

Trimble Navigation Limited 2002, [Mapping Systems: General reference](#), viewed 9 September 2015, p62.

USGS 2006, [Map Projections](#), viewed 14 August 2015.

ICSM 2 April 2001, [What is the difference between WGS84 and GDA94?](#), viewed 8 September 2015.

Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection

Version: February 2018

8. Operating a basic handheld Global Positioning System unit for an investigation or compliance inspection

8.1. Purpose and scope

This document describes the method for sound locational data collection using a basic handheld Global Positioning System (GPS) unit for an investigation or compliance inspection. It does not provide details for using a differential or relative positioning GPS.

GPS is the United States of America (USA) system of global positioning and is the most commonly used way of describing positioning systems. However, in reality a number of positioning systems are now used such as GLONASS (Russian satellite system) and Galileo (European satellite system) and the more appropriate term is Global Navigation Satellite System (GNSS). In this document the term GPS is used to encompass all systems, not just the USA global positioning system.

8.2. Associated documents

Sampling design and preparation: Background information on the Global Positioning System and handheld units

8.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

8.4. Permits and approvals

No permit is required to carry out the procedures within this document.

8.5. Skills, training and experience

No skills, training or experience is required to conduct the procedure within this section.

8.6. Equipment

Equipment should include:

- handheld GPS unit or a smart phone, iPad or tablet as a primary device if it has been set up appropriately (e.g. with specific software to record positions)
- a smart phone, iPad and tablet may be used as a secondary device to confirm readings
- GPS device manual.

8.7. Procedure

8.7.1. Prior to undertaking fieldwork

8.7.1.1. Pre-field trip check of GPS

1. Check the GPS settings to ensure standard settings are used. Recommended settings are outlined in
2. Table 4. Record settings in your notebook.
3. Check the time and date on the GPS against your computer or smartphone.
4. Make sure GPS unit has sufficient memory for all new waypoints and tracks.

Note:

- It is good practice to regularly synchronise your smartphone clock with broadcast time services on TV or radio. If time is critical to your observations use a designated time service such as the VNG radio service

(http://tufi.alphalink.com.au/time/nsc_vng_leaflet.pdf).

- If using points in open water it is recommended that two GPS units are used to confirm the readings as there are no land marks to check positions against. Use a smart phone or tablet with GPS as a third method of checking position if both handheld GPS units show coordinates that are significantly different to each other.

Table 4: GPS unit settings

Setting with options	Select option
Units	Metric
Datum	GDA 94 (Geodetic Datum of Australia)
Location Format	UTM UPS
North Reference	True

8.7.1.2.

Pre-field 'calibration' of GPS

1. Find a datum permanent survey mark that is accessible both before and after sampling, preferably close to the investigation area. Permanent survey marks (PSM) in the area of interest can be located using the Google Earth™ and Queensland Globe (<https://data.qld.gov.au/maps-geospatial/qld-globe>). Load the Location Globe to find details of PSM in the area. Download the survey control report (SCR) for the PSM to be used by clicking on the PSM in the globe.
2. Go to the permanent survey mark, turn GPS on and place GPS next to the survey mark.
3. Turn on the GPS and allow time for the GPS to initialise.
4. Ensure that the GPS is set to calculate 3D positions, and that enough time has elapsed for the GPS to acquire enough satellites to provide a 3D position.
5. Take a photo showing both the permanent survey mark and the GPS next to it (**Figure 6**).
6. Take a photo of the GPS's serial number with the permanent survey mark.
7. Record the:
 - date, time and location (e.g. street and town) of the calibration
 - PSM number
 - GPS reading
 - GPS serial number or make and model of the GPS
 - the uncertainty reading on the screen.
8. Note that photos were taken.
9. If working in pairs, note the name of the corroborator who also witnessed the testing process.
10. Compare GPS reading to the coordinates in the SCR for the PSM being used. Check that the difference between the coordinates on the GPS and coordinates of the survey mark are within the accuracy defined for the GPS unit in the user manual for that unit. An example is given below (Example 1).

Note: Having UTM UPS selected as the location format on the GPS makes this comparison easier, as each easting or northing equates to meters (e.g. 503993.422 m E is 2 m from 503995.422 m E). If using latitude and longitude, record values to the nearest point of a second (i.e. 27° 29' 42.6" and 153° 01' 47.7").



Figure 6: GPS next to permanent survey mark 55309

Example 1: Comparison between GPS and survey control report (SCR) for the permanent survey mark (PSM)

- The SCR states the PSM is at:
56 J 503993.422 m E / 6958558.367 m N
- The GPS manual states the accuracy of the GPS is to 10m.
- Therefore the GPS can be within a radius of 10m of the SCR coordinates.

8.7.1.3. Reference points

When investigating a remote site that may be difficult to return to, it is advisable to collect GPS location data at permanent landmarks that are visible on remotely sensed imagery in the area of investigation to allow remotely sensed imagery to be georeferenced after the field inspection has been completed.

Prior to undertaking the investigation, use remote imagery (such as recent aerial photographs or Google Earth™) to select approximately 10 reference points evenly spread over the investigation area. Not all 10 points need to be surveyed, but it is recommended 10 are chosen initially in case it is not possible to access all points. Ideally five reference points will be recorded during a field inspection. The reference points need to be:

- permanent (not mobile)
- in a clear area
- not underneath high voltage power lines
- where two obvious features meet, e.g. where two roads cross, where a road crosses a creek at a bridge, where two fenced boundaries meet (intersections of fences also serve to provide a control linkage to the cadastre and may be used to upgrade the accuracy of the DCDB if property boundary definition is important to your study)
- clearly identified on the remote imagery (aerial photograph or Google Earth™ image).

These localities and their GPS reference locality should be described in a field notebook so that they can be recalled later.

8.7.2. Undertaking fieldwork

8.7.2.1. Reference points

1. If reference points are to be collected, take a GPS reading at each of the reference points prior to attending the investigation site using the method in Section 7.1.3.
2. After leaving the investigation site, repeat readings (if possible) to obtain a duplicate reading at each reference point.

8.7.2.2. Recording coordinates

1. Upon reaching the site, where possible choose a location where the sky can clearly be seen to take the reading.
2. Satellite signals can be weakened by vegetation cover, water bodies, metal surfaces (e.g. vehicles), glass (including vehicle windscreens), buildings and similar objects. Hold the GPS away from your body.
3. Ensure the reading is not taken underneath high voltage power lines as power lines create magnetic fields and can cause destructive interference to satellite signals passing through the magnetic field.
4. Hold GPS stationary for at least 30 seconds. Before taking readings, ensure that the GPS is able to calculate a 3D position. It is recommended that you do not collect 2D positions because of the problem encountered with the altitude measurement. When you set the receiver to calculate 2D positions, you are replacing one satellite measurement (altitude) with a fixed measurement. If this altitude is incorrect, the latitude and longitude will also be incorrect.
5. It is recommended that the coordinates in the GPS unit are saved as a waypoint. Name the waypoint appropriately so it can be identified later and use this same name in the field notes.
6. Record the coordinates of this site in the field notebook and/or take photo of coordinates on the screen. Record relevant features in a notebook.
7. Take photographs of the site as a visual confirmation of the site and record photo directions in a field notebook. It is recommended that they are taken in:
 - four directions - downstream, right shore, upstream, left shore are photographed if surveying an aquatic environment (rotating in a clockwise direction)
 - eight directions - N, NE, E, SE, S, SW, W, NW, so that photographs all have side-lap and can be joined in a panorama of the site for terrestrial systems.. Always include a hat/person/bag in the first photo (north) and always rotate in a clockwise direction looking down at your point.
8. It is recommended that you turn your GPS unit off and on again, cross check the GPS readings against a secondary unit. It may be useful to intermittently cross check with a secondary unit throughout the day.

Note: Many GPS units include an electronic compass. In some GPS models the compass reading is only accurate whilst you are moving. When stationary, and if you change your orientation (direction), the GPS unit compass may not reflect a correct directional reading until you begin to move again.

8.7.2.3. Post-field 'calibration' of GPS

Before returning to the office, go back to the permanent survey mark used in the original pre-field 'calibration'

Follow steps outlined in section 8.7.1.2.

8.7.3. Upon return to the office

8.7.3.1. Data handling

1. Download waypoints and track log files from the GPS unit and store securely as an unaltered clearly labelled master copy. Preferably on CD or DVD.
2. Create a working copy for use in GIS applications.

Choosing a laboratory and analytical method, holding times and preservation

Version: February 2018

9. Choosing a laboratory and analytical method, holding times and preservation

9.1. Purpose and scope

This document outlines the requirements involved in selecting a laboratory and appropriate analytical methods for a sampling program and associated holding times and preservation of samples.

9.2. Associated documents

Sampling design and preparation:

- *Preparation for sampling*
- *Sampling scope and design*

9.3. Selection of laboratory

Select laboratories that demonstrate Good Laboratory Practice (GLP) and perform according to the standard AS ISO/IEC 17025-2005 (R2016): General requirements for the competence of testing and calibration laboratories. This is usually demonstrated by laboratories holding accreditation with the National Association of Testing Authorities (NATA). Although NATA accredited laboratories are recommended, on occasion exceptional circumstances arise where specialist analysis and interpretation may be required. If the analysis is not available through using a NATA accredited laboratory, select another laboratory that holds the required expertise. It is important to validate the procedure including its detection or reporting limits and accuracy.

Analysis of samples taken for statutory purposes under the Environmental Protection Act 1994 and its subordinate legislation, including the Environmental Protection Regulation 2008 and the Environmental Protection (Water) Policy 2009 should be undertaken by NATA accredited laboratories.

9.4. Selection of analytical methods

The laboratory should be consulted prior to sampling in regards to the appropriate analytical method that will provide the required information to meet the purpose of the sampling program. The analytical methods selected may well dictate the sampling procedures required.

The method should be:

- appropriate for the type of sample and for the expected concentration range of the analyte to be measured
- verified or validated (e.g. by proficiency tests, using certified reference materials or standard addition techniques) (see NATA Technical Note No. 17; NATA 2013)
- accredited by NATA, or at least equivalent to (e.g. at least national accreditation if samples sent overseas).

Note: Laboratories receive NATA accreditation to perform specified analytical methods. Always check the laboratory has accreditation or validation for the required methods.

If the laboratory is not NATA accredited, samples should be analysed in accordance with a method specified in one of the following reference texts:

- *APHA AWWA Standard Methods for the Examination of Water and Wastewater (current version)*
- *USEPA Clean Water Act Analytical Methods (current version)*
- *ASTM, Annual Book of ASTM Standards Volume 11.01 Water (I), 11.02 Water (II) (current version)*
- relevant Australian Standards (current version)
- relevant ISO Standard (current version).

Alternative methods to those described in the reference texts can be used, provided the laboratory has validated the alternative method and proven that the results obtained are equivalent to those that would be obtained using the prescribed method. The laboratory also needs to show that the results are within the uncertainty stated for the prescribed method.

9.5. Limits of detection and reporting

The limit of reporting (LOR) for an analytical method should be lower than the benchmark (such as water quality objective, water quality guidelines or trigger values, Environmental Authority (EA) limits or other standards) to which the results will be compared. The LOR is the lowest concentration of an analytical parameter that can be detected by a particular method that has acceptable precision and accuracy. If the LOR is higher than the benchmark, alternative methods of sampling and/or analysis should be investigated (e.g. passive sampling).

9.6. Holding times and preservation

Holding times (how long a sample can be kept between collection and analysis), and preservation of samples is critically important for the integrity of a sampling program, as the constituents of samples may change between collection of samples and analysis (as a result of chemical, biological or physical reactions). (AS/NZS 5667.1:1998). Guidance for different holding times and preservation methods is provided in:

- *APHA AWWA Standard Methods for the Examination of Water and Wastewater (current version)*
- *USEPA Clean Water Act Analytical Methods (current version)*
- *Australian and New Zealand Standard: AS/NZS 5667.1:1998.*

Ideally the holding times and preservation methods specified in the aforementioned documents should be adhered to. However alternative methods may be determined for samples collected as part of an on-going monitoring program (e.g. Oudyn et al. 2012) or say if a preservative is considered hazardous. Alternative methods can be used in such cases, if the alternative method has been validated and the laboratory can demonstrate the results obtained are equivalent to the results that would be obtained using the relevant prescribed holding times/preservation method.

The analytical laboratory should be contacted for advice on holding times and preservation before any samples are collected.

Some holding times are extremely short (for example pH and chlorine) and the use of in situ tests and test kits may be more appropriate than laboratory analysis.

9.7. References and additional reading

APHA AWWA (current version) Standard Methods for the Examination of Water and Wastewater, American Public Health Association (APHA), the American Water Works Association (AWWA) and the Water Environment Federation (WEF).

ASTM (current version), Annual Book of ASTM Standards: Volume 11.01 Water (I), 11.02 Water (II).

Australian and New Zealand Standard (AS/NZS) 5667.1:1998, Water quality—Sampling Part 1: Guidance on the design of sampling programs, sampling techniques and the preservation and handling of samples.

National Association of Testing Authorities, Australia 2014, Guidelines for the validation and verification of quantitative and qualitative test methods Technical Note 17—October 2013, viewed 8 December 2016, http://www.nata.com.au/nata/phocadownload/publications/Guidance_information/tech-notes-information-papers/technical_note_17.pdf.

Oudyn, F, Lyons, DJ, Pringle, MJ 2012, 'Appropriate maximum holding times for analysis of total suspended solids concentration in water samples taken from open-channel waterways', *Water Science and Technology*, 66, 1310-1316.

USEPA (current version), Clean Water Act Analytical Methods, US Environmental Protection Agency, Available from: <https://www.epa.gov/cwa-methods>.

Section B: Physical and Chemical Assessment

Background information on water quality measurements using in situ water quality instruments

Version: February 2018

1. Background information on water quality measurements using in situ water quality instruments

1.1. Purpose and scope

This document provides background information on in situ water quality measurements prior to undertaking water quality sampling.

1.2. Associated documents

Physical and chemical assessment:

- *Water quality sampling using in situ water quality instruments*
- *Chlorophyll a sample collection methods*
- *In situ water quality sampling using a Secchi disc*

1.3. Introduction

In situ water quality sampling is the measurement of physical and chemical parameters in a water body at the time of sampling. This is usually done because the measured parameters change rapidly (for example temperature). The data is as valid as data measured in a laboratory provided the field instruments are calibrated. In situ data are often required to aid the interpretation of other water quality results.

The most common method of measuring in situ water quality is with a multi-parameter water quality instrument (**Figure 7**). The sonde of a multi-parameter water quality instrument is a collection probes that measure individual parameters. Whilst the configurations of probes vary with each instrument, the most common are dissolved oxygen (DO), temperature, pH, electrical conductivity (EC), turbidity and depth. Probes are available that measure other parameters such as chlorophyll, oxidation reduction potential (ORP), ammonia, ammonium, nitrate and chloride. Single parameter instruments are also available (e.g. a pH meter).

Probes that measure nutrients and chlorophyll must be regularly checked against laboratory analysed samples. Chlorophyll probes, in particular, may only provide relative concentrations of chlorophyll in a water column, and need to be calibrated with chlorophyll in water samples collected on the same field trip.

1.4. Overview of measurements

1.4.1. Temperature

Accurate temperature measurements are required for accurate determinations of pH, specific electrical conductivity, and dissolved oxygen. Stratification is common in summer months when surface waters are much warmer than bottom waters. Accordingly, unless the water is shallow (less than 0.5m) and flowing, take temperature readings at different (measured) depths (along with other parameters) in order to define the stratification (if present). Use markings on meter cable if depth sensors are not available on your equipment.

Warm water is less capable of retaining dissolved oxygen than cold water. For this reason, temperature should be measured at the same place in the waterbody where dissolved oxygen is measured. This ensures the resulting data relate to the same body of water at the same time.

The toxicity of ammonia and cyanide changes depending on temperature. Therefore temperature must always be measured when monitoring ammonia and cyanide.



Figure 7: Testing waters with a multi-parameter water quality meter

1.4.2. pH

The pH measures the acidity or alkalinity of water, with a pH of 1 being strongly acidic, a pH of 7 being neutral, and a pH of 14 being strongly basic (or alkaline). Generally, the pH of fresh surface waters are between 6.5 and 8.0, and the pH of most marine waters is close to 8.2 (ANZECC and ARMCANZ 2000). Marine water generally has a stable pH as the high concentrations of dissolved carbonates provide a high buffering capacity (resistance to pH change) by neutralising any hydrogen ions (from acid). There are many processes (natural or human induced) that may elevate or decrease pH of water. For example, acid rock drainage (natural or human induced) or acid sulfate soils can decrease the pH of a water body to 2, or an algal bloom can increase pH readings to 9.5.

The pH changes the toxicity of ammonia, aluminium and cyanide, and must be measured at the same time and location when analysing for these chemicals.

1.4.3. Dissolved oxygen (DO)

Dissolved oxygen (DO) are reported in units of milligrams of oxygen gas (O₂) dissolved in each litre of water (i.e. mg/L) or as a percentage of the maximum amount of DO that is possible in a waterbody at a specified temperature and salinity (% saturation). Most multi-parameter water quality instruments containing DO sensors compensate automatically for temperature and salinity when calculating DO saturation (verify by checking the user manual for the instrument). DO concentrations are dependent on atmospheric pressure, and this is taken into account during instrument calibration.

Considerable differences between DO concentrations at the surface and at depth in waterbodies can result from stratification of the water column, due to temperature or salinity effects. This effect is usually most pronounced in summer months when surface waters are considerably warmer than deeper waters.

Degradation of a natural waterway, by interference in the natural flow and/or the build-up of excessive nutrients, can cause the development of stagnant conditions and excessive growth of aquatic plants and/or algae. Under natural conditions with high algal density during daylight, super-saturation (more than 100 per cent DO) can occur. Excess DO can lead to 'gas bubble disease' in fish, where oxygen bubbles can form in the vascular system, gill lamellae and eyes, amongst other organs, which can lead to death. In addition, where algal and plant growth is excessive (as indicated by high DO reading), algal and plant respiration at night can deplete the available dissolved oxygen sufficiently to result in a fish kill. Therefore, the time of day dissolved oxygen is collected can be important for interpretation of data. It is preferable (if possible) that DO readings are taken very early in the morning when the lowest dissolved oxygen levels will be present. DO at concentrations of less than 2 mg/L can be associated with fish kills. Low DO measurements are also often caused by the introduction of large loads of organic matter into

waterways at the start of the wet season or due to organic matter from sewage spills.

The Queensland Water Quality Guidelines (DES 2009) present DO measurements as % saturation whereas DO measurements associated with fish kills tend to be presented as mg/L, and therefore both measures (mg/L and % saturation) should be recorded.

1.4.4. Electrical conductivity (EC)

Electrical conductivity (EC), often simply called conductivity, is a measure of the ability of water to conduct an electrical current. The ability to conduct an electric current is due to the presence of dissolved salts. Thus, EC is used to calculate salinity and the concentration of dissolved salts in a waterbody. The formal unit for conductivity is siemens per metre (S/m), however microsiemens per centimetre ($\mu\text{S}/\text{cm}$) is more commonly used when measuring fresh or brackish waters, and millisiemens per centimetre (mS/cm) when measuring estuarine and marine waters.

EC varies with temperature, and values reported are usually corrected to 25°C. Such data are known as Specific Conductance. A difference of 5°C can alter conductivity by approximately 10%. Many conductivity instruments have compensation functions so that EC at 25°C can be read directly (verify by checking the instruments manual). However, if the meter does not automatically compensate for the temperature, a manual correction can be made by using the formula:

$$K_{25} = \frac{K_t}{1 + 0.019(t-25)}$$

where:

K25 = corrected (25°C) electrical conductivity of the water (Specific Conductance)

Kt = electrical conductivity at the measured temperature (t°C)

t = water temperature (°C) where and when electrical conductivity is measured

1.4.5. Salinity

Salinity is the measure of the dissolved salt content of a body of water. Salinity in parts per thousand (g/L) can be calculated from conductivity at 25°C using the formula:

$$S = a_1(K_{25}) + a_2(K_{25})^2 + a_3(K_{25})^3 + a_4(K_{25})^4 + a_5(K_{25})^5 + a_6(K_{25})^6$$

where:

S = salinity (in g/kg, % or parts per thousand (‰))

K25 = electrical conductivity of the water at 25°C (in mS/cm)

a1 = 4.98 x 10⁻¹

a2 = 9.54 x 10⁻³

a3 = -3.941 x 10⁻⁴

a4 = 1.092 x 10⁻⁵

a5 = -1.559 x 10⁻⁷

a6 = 8.789 x 10⁻¹⁰

1.4.6. Total dissolved solids

Total dissolved solids (TDS) include total dissolved salts but also non-ionised species (e.g. sugars, other organics and colloidal particles). Therefore, TDS values are often larger than total dissolved salt values for the same water sample.

Total dissolved solids are either determined by:

- filtering a water sample, evaporating a weighed amount of filtrate to dryness in a weighed dish, drying to a constant weight, and determining the increased mass of the dish, or
- calculating the approximate TDS (for typical fresh waters) from conductivity using the formula:

$$\text{TDS} = \text{MF} \times K_{25}$$

where:

TDS = Total Dissolved Solids (mg/L)

MF = multiplication factor (0.64 for drinking water⁴, 0.67 for livestock drinking water⁵)

K₂₅ = EC of the water at 25°C (mS/cm)

1.4.7. Turbidity

The turbidity of a water body is a measure of the presence of soluble, suspended and colloidal particles that hinder the transmission of light through water. Turbidity can potentially affect the rate of photosynthesis, and therefore the growth of plants or algae in the water body. Turbidity can be measured directly using probes, and is typically expressed using Nephelometric Turbidity Units (NTU).

1.4.8. Transparency

Transparency is a measure of how far light can pass through water. In waterways this translates to how deeply sunlight penetrates through the water. The degree of transparency at any given depth of water affects the rate of photosynthesis, and hence the growth of coral, plants or algae in the water body. Transparency can be measured using the Secchi disc (see In situ water quality sampling using a Secchi disc document). A Secchi disc has the advantage over a single turbidity reading as it integrates turbidity over depth (where variable turbidity layers are present).

1.5. References and additional reading

ANZECC and ARMCANZ 2000, Australian and New Zealand guidelines for fresh and marine water quality, Volume 2, Aquatic ecosystems, Australian and New Zealand Environment and Conservation Council, Agriculture and Resource Management Council of Australia and New Zealand.

DEHP (Department of Environment and Heritage Protection) 2009, Queensland Water Quality Guidelines, Version 3, ISBN 978-0-9806986-0-2.

NHMRC and NRMCC (National Health and Medical Research Council and National Resource Management Ministerial Council) 2011, Australian drinking water guidelines, National Water Quality Management Strategy, Paper 6. NHMRC and NRMCC, Commonwealth of Australia, Canberra. Available from: www.nhmrc.gov.au/guidelines/publications/eh52.

⁴ Australian drinking water guidelines (NHMRC and NRMCC 2011)

⁵ Australian and New Zealand guidelines for fresh and marine water quality (ANZECC and ARMCANZ 2000)

Water quality sampling using in situ water quality instruments

Version: February 2018

2. Water quality sampling using in situ water quality instruments

2.1. Purpose and scope

This document describes the correct method for the use of in situ water quality instruments.

2.2. Associated documents

Physical and chemical assessment:

- Background information on water quality measurements using in situ water quality instruments
- In situ water quality sampling using a Secchi disc

Sampling design and preparation:

- Permits and approvals
- Record keeping including taking field photographs and videos

2.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

2.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

2.5. Skills, training and experience

Staff using this method should have previous training and experience in the use and calibration of the equipment described.

2.6. Field equipment

Equipment specific to this method includes:

- single or multi-parameter water quality instrument
- data sheets, notebook or field computer
- waders, gumboots, lifejacket or similar personal protective equipment (shore based sampling).

2.7. Procedure

2.7.1. Instrument maintenance and calibration

Due to the variety of water quality instruments available, it is not practical to provide instrument-specific advice on storage, calibration and maintenance in this document. Before taking an instrument into the field, you should be familiar with the contents of the operating manual for that specific instrument, and ensure that it is stored, calibrated, maintained and used as per manufacturer's instructions. Detailed records of calibration and maintenance must be kept.

2.7.1.1. Records

For each instrument the following procedures and documents should be established and kept up to date:

- list of spare parts and suppliers/sources of repair
- written inspection, maintenance and calibration schedules
- log book which has a record of inspection, maintenance and repair activities detailing dates and people involved
- log book which has a record of calibration activities detailing dates, times, results, standard(s) used and people involved.

Keeping such records allows it to be determined whether the equipment has been maintained in a sound operating condition and the recorded data are credible. This information is vital when undertaking investigations.

2.7.1.2. Calibration

Calibration must be undertaken as per the instrument manufacturer's instructions. Some general issues to consider are:

- adequate supplies of calibration standards should be available. Make sure the standards have not expired. Standards can be reused provided the probes are rinsed in distilled water and dried thoroughly between standards, as to not contaminate them or dilute them.
- the calibration should be conducted using standards in the range of values expected to be encountered in the field. This is particularly important when calibrating electrical conductivity for use in fresh, tidal or marine waters
- if manufacturer's procedures does not refer to temperature calibration, manual temperature readings taken using a thermometer should be compared to the instrument temperature readings
- the calibration must be performed and recorded before the start of a field trip, and should be checked at the conclusion of each field trip.
- it is advisable to re-check calibration daily during an extended period of field use. These in-field checks should be recorded in a notebook and later transcribed into the calibration logbook for the instrument.
- variability in performance shown by calibration checks should be reported with the data, and provided to the instrument servicing agent.

Because laboratory calibrations rely on buffered solutions, a cross check comparison (shadow testing) of multiple instruments using an environmental water sample should be conducted from time to time. This is done using two or more instruments simultaneously to test a bucket of typical environmental water (e.g. from a creek). The comparison of readings should be within the range of the stated accuracy specifications of the parameter (refer to the operating manual of the instrument). If only one instrument is available and it is not possible to undertake shadow testing, it is recommended to return it to the manufacturer for regular calibration (as per the manufacturer's instructions).

2.7.2. Taking a reading with a multi-parameter water quality meter

2.7.2.1. Shore based procedure

1. One person holds the water quality meter on the shore, whilst another takes the sonde and wades out into the water body.
2. Move to the centre of the water body if possible. If water is flowing face so the current is flowing towards you.
3. Allow any water disturbed by your movement to the site to flow past you.
4. Turn the meter on and place the sonde in the water to a depth of 0.2m (to reduce impact surface slicks) upstream from where you are standing if there is flow, or beyond the area of disturbance if there is not flow.
5. Allow the readings to stabilise.
6. Follow the manufacturer's direction for taking the readings.
7. Record the measured values in a notebook and save the reading if a memory function is available.

8. Where the water is deep enough, repeat steps 5-7 at 0.5m to 1m intervals (depending on depth) until the bottom is reached (to provide a depth profile).
9. Record the following details:
 - site details (e.g. site code, site name, waterway, GPS co-ordinates)
 - date and time of measurement
 - all measured values
 - any factors that may have affected the measurement (e.g. presence of an algal bloom, recent rainfall etc.).

Note: If it is not safe to enter the water (e.g. due to fast flowing current or potential presence of dangerous fauna such as crocodiles) or if there is no flow and the sediment is disturbed by the sampler wading into the water, the procedure may be modified to lower the sonde into the water from a river bank, bridge or boat.

2.7.2.2. Boat based procedure

1. Whilst the boat is moving forward, switch off the engine and allow the boat to drift forward and clear of possible contamination caused by the motor or propeller.
2. Turn the meter on and place the sonde in the water to a depth of 0.2m (to reduce impact surface slicks).
3. Follow the manufacturer's direction for taking the readings.
4. Allow the readings to stabilise.
5. Record the measured values in a notebook, and save the reading if a memory function is available.
6. Repeat steps 5-7 at 2m intervals in estuarine water (1m intervals in freshwater) until the 0.5m off bottom is reached (to provide a depth profile). It is usually possible to take an extra reading between the deepest two-meter interval and the bottom. This is taken at the closest 0.5m interval from the bottom.
7. Record the following details:
 - site details (e.g. site code, site name, waterway, GPS co-ordinates)
 - date and time of measurement
 - all measured values
 - any factor that may have affected the measurement (e.g. presence of an algal bloom, recent rainfall etc.).

Note: Generally, if sampling in a tidal area, sampling is to be conducted on the ebbing (outgoing) tide.

2.7.2.3. On return to the office

Download data from water quality meter (always keep a copy of unmodified data files on a backed up drive).

Store notebook or field sheets in a safe place.

2.7.3. General considerations

- Sensors on sondes should be kept moist or wet at all times (depending on the sensor), and not be allowed to dry out. See the manufacturer's manual for specific directions on storing and transporting your instrument. Distilled water should not be used.
- Between sites, it can be advantageous to sit the sonde in water from the previous site to maintain sensor stability.
- Do not allow the sonde to touch the substrate as there is a risk of damage to the sensors from sticks, rocks, debris and anoxic sediments. Touching the bottom can also stir up sediments into the water column, changing the natural characteristics of the water being sampled. Depth can be determined in a boat by using the depth sounder. Thoroughly clean the sonde if it has accidentally made contact with the substrate before taking any further readings.
- Stratification with the water column can occur for a number of parameters, therefore depth profiling is necessary in waterbodies deeper than 0.5m.
- Verify that the instrument you are using compensates for factors such as temperature when measuring electrical conductivity, or whether results need to be adjusted by calculation (see Background information on water quality measurements using in situ water quality instruments document).
- Under natural conditions such as high algal density during sunlight, it is possible to have dissolved oxygen (DO) super-saturation (more than 100% DO).

2.7.4. Atypical instrument readings

Atypical readings may be due to abnormal conditions in a waterbody, but they could also be a sign of problems with the equipment or equipment failure. Typical readings for dissolved oxygen, electrical conductivity and pH in potable (drinking), fresh and marine water are presented in **Table 5**. If readings appear atypical, the first step should be to check for equipment problems, such as a broken electrical cable or insulation, fouled sensor or faulty probe, depleted batteries, etc. If the equipment appears sound take extra measurements to confirm that the results are valid (i.e. move to another site) and check the calibration.

Table 5: Typical ranges for dissolved oxygen, conductivity and pH in different water types

Parameter	Potable water	Fresh water	Marine water
Dissolved oxygen (DO)		Typical concentrations under ambient conditions 6–10mg/L. Values may be higher if algal blooms are present, or lower if anoxic conditions are present.	
Electrical conductivity (EC)	50–500 μ S/cm	<1500 μ S/cm	~52 000 μ S/cm
pH		6–8.5. These may be lower if acid rock drainage or acid sulfate soils/sediments are present. The total range of pH values is generally 0–14.	

In situ water quality sampling using a Secchi disc

Version: February 2018

3. In situ water quality sampling using a Secchi disc

3.1. Purpose and scope

This document describes the method for the correct use of the Secchi disc for the measurement of transparency in surface water bodies.

3.2. Associated documents

Physical and chemical assessment:

- *Background information on water quality measurements using in situ water quality instruments*
- *Water quality sampling using in situ water quality instruments*

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping including the taking of field photographs and videos*

3.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

3.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

3.5. Skills, training and experience

No skills, training or experience is required to conduct the procedure within this section.

3.6. Equipment

See Appendix 1 for example equipment checklist.

3.7. Procedure

Note:

- A Secchi disc is not appropriate for use in shallow waters where the disc can still be seen when resting on the bottom of the water body.
- If prescription glasses or contact lenses are normally worn, these should be worn when undertaking Secchi depth measurements. Tinted lenses or sunglasses should not be worn, as they could affect the depth at which the disc is no longer visible and re-appears.
- Weather conditions can affect the visibility of the disc through the water (i.e. cloud cover and wind).
 1. Check Secchi disc (**Figure 8**) for damage and untangle any knots in the rope.
 2. Lower the Secchi disc into the water on the sunny side of the vessel (**Figure 9**) until the black/white interface is no longer visible.
 3. Record this depth as D1 (measurement should be made at a precision of half the distance between marks on rope).
 4. From this point raise the disc until the black/white interface of the Secchi disk reappears.
 5. Record this depth as D2.
 6. Calculate Secchi depth using the following formula:

$$\text{Secchi depth} = \frac{D1 + D2}{2}$$

7. Record the following details:
 - site details (e.g. site code, site name, waterway, GPS co-ordinates)
 - date and time of measurement
 - distance between observers eyes and water level
 - D1, D2 and the calculated Secchi depth
 - any factor that may have affected the visibility of the disc (e.g. wind disturbing the surface of the water).
8. Readings should be taken between two hours after sunrise and two hours before sunset.



Figure 8: Secchi disc attached to 3kg weight and tether



Figure 9: Secchi disc being lowered into the water

3.8. References and additional reading

Australian Standards 1993 (R2017), Waters–Part 7: The construction and use of the Secchi disc, AS 3550.7-1993 Homebush, NSW.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Data sheets, notebook or field computer	
GPS, phone, camera, Go Pro or body cam	
300mm disc painted with quadrants alternating in flat black and flat white in waterproof paints	
3kg weight, suspended below the centre of the disc with an eye bolt	
Tether of non-stretch nylon rope (surveyors Kinlon poly-chain) with waterproof measurement graduations, attached to eye bolt on the Secchi disc	

Manual collection of surface water samples (including field filtration)

Version: June 2018

4. Manual collection of surface water samples (including field filtration)

4.1. Purpose and scope

This document describes the method for manually collecting water quality samples.

4.2. Associated documents

Physical and chemical assessment:

- *Water quality sampling using in situ water quality instruments*
- *In situ water quality sampling using a Secchi disc*

Sampling design and preparation:

- *Quality control for water and sediment sampling*
- *Preparation for sampling*
- *Permits and approvals*
- *Record keeping including the taking of field photographs and videos*

4.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

4.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

4.5. Skills, training and experience

Skills, training and or experience required to understand and or undertake this method include experience in using the equipment described in the method.

4.6. Equipment

See Appendix 1 for example equipment checklist.

4.7. Procedure

4.7.1. Overview

Samples should be collected directly into the laboratory supplied containers when sampling waters where possible, as this will reduce the risk of contamination. Direct sample collection is the preferred procedure if the environment is safe (e.g. during low flow conditions), and sample bottles do not contain preservative.

Where there is no flow (e.g. in dams and isolated pools) samples may be collected using an extendable sampling pole (**Figure 10**), rather than wading in, so as not to disturb the substrate. However, this may not represent the bulk of the waterbody as it is an edge sample. An alternative method is to move slowly into the middle of the water body (if safe) and collect the sample away from any plume caused by movement into the water body. The choice of method will depend on the objectives of the sampling.

During high flow conditions or where entry into the water is not possible (due to the presence of crocodiles or chemical contamination), the use of an extendable sampling pole or water sampler (such as a Niskin bottle or Van Dorn sampler (**Figure 11**)) are recommended. Water samplers can be used to collect samples at discrete depths. The choice of water sampler will depend on the sampling program. For example, if water samples are being collected for ultra-trace metal analysis, it may be appropriate to obtain a water sampler that has a Teflon-coated internal mechanism. Contact your scientific supply company for more information.



Figure 10: An extendable sampling pole



Figure 11: Van Dorn sampler

4.7.2. Preparation for sampling

1. Prior to leaving for the field, inspect all equipment and ensure it is in good working order and has been cleaned appropriately. Make sure if you are using an intermediate container that is appropriate for the analytes/s being sampled for.
2. On reaching the sampling site, prepare a clean work area and ensure all equipment that you will require is unpacked and easily accessible.
3. Pre-label all sample containers if possible.
4. Confirm the order that sample containers will be filled. When preparing for sampling note whether:
 - the sample container requires rinsing
 - the sample container contains preservative
 - the sample requires 'no head space'.

Note:

- If the sample container contains preservative, it will not be possible to collect water directly into the sample bottle. It is recommended that a clean laboratory supplied bottle (made of the same material, washed appropriately and without the preservative present) be used to collect the sample, with subsequent transfer to the bottle with preservative. This clean collection bottle should not be used at more than one site.
- Check with your laboratory on how much sample is needed for analysis. This is particularly the case for field filtered samples in turbid waters, where it may be difficult to filter. .
- If undertaking ultra-trace sampling, double bag samples to protect them from cross contamination from leakages and melted ice.

4.7.3. Collecting samples directly from a stream or river

1. Label sample containers.
2. Identify a representative area to collect the water sample and determine an appropriate and safe access route that will minimise the risk of disturbance of the substrate.
3. Collect in situ water quality measurements before collecting water samples for chemical analysis. Measurement of temperature, dissolved oxygen and conductivity can indicate the presence of stratification in the water, and may determine where in the water column samples should be collected. See Water quality sampling using in situ water quality instruments document.
4. Put on powder free gloves immediately prior to collection of the water sample. Gloves must be stored in a clean environment (e.g. in a plastic bag).
5. To collect the sample, wade into the mid-point of the stream or river, and face into the direction of the flow. This is so any potential contamination from substrate disturbance will flow away from where the sample is being collected.
6. Remove the lid of the bottle, ensuring your fingers do not come into contact with the internal surfaces of the sample container or lid.

7. Invert the sample container fully and submerge to a depth of 0.3m below the water surface—avoid surface scums and debris in the water including macrophytes. If the water is less than 0.6m deep, the sample should be collected at mid-water column.
8. Rotate the sample container into the direction of flow.
9. If rinsing the bottle, allow the sample container to fill at least one third of the container volume. Remove from the water and recap.
10. Shake the sample container gently and pour the water downstream of the sample collection point.
11. Complete the rinse procedure three times.
12. To fill the sample container, repeat steps 6 to 8 and then allow the sample container to fill completely.
13. Recap the sample container.
14. Return to the shore and check that the details on the sample container are correct.
15. Place the sample container in a cooler box (with ice or ice bricks) or refrigerator and chill. Double bag samples if ice is to be used.
16. Fill out the chain of custody form.

Note:

If sample is to be collected from a boat, collect the sample as close to the front of the boat as practicable, as it is moving slowly forward into the current. Be aware of any potential contamination from the boat.

If undertaking analysis for ultra-trace metals it is recommended that USEPA *Method 1669 Sampling ambient water for trace metals at EPA water quality criteria levels* (USEPA 1996) be consulted.

4.7.4. Sample collection with an extendable sampling pole

Extendable sampling poles (see **Figure 10**) should ideally have an attached adjustable bottle holder that allows laboratory supplied bottles to be fitted directly to the pole. This eliminates the need for an intermediate sample container and reduces the risk of sample contamination. The bracket should angle the mouth of the bottle away from the pole, thereby reducing the potential for water to drip off the pole into the sample container. The use of rope to hold clasp brackets to the pole should also be avoided to minimise the potential for water to drip off the rope and bracket into the sample container.

The procedure for collection using an extendable sampling pole is:

1. Collect in situ water quality measurements before collecting water samples for chemical analysis. Measurement of temperature, dissolved oxygen and conductivity can indicate the presence of stratification in the water, and may dictate where in the water column samples should be collected. See *Water quality sampling using in situ water quality instruments* document.
2. Clean extendable pole and adjustable bottle holder thoroughly prior to the collection of samples. Equipment should be cleaned with phosphate-free laboratory grade detergent and rinsed with clean water.
3. Extend the sampling pole to the required length and check that all surfaces have been cleaned.
4. Place the sample container into the adjustable bottle holder.
5. Remove the lid of the sample container ensuring your fingers do not come into contact with the internal surfaces of the sample container or lid. Do not place the sample lid on the ground.
6. Move to the edge of the water and invert the sample container and submerge to a depth of 0.3m below the water surface. Avoid surface scums and debris in the water. Avoid sampling over macrophyte beds.
7. Rotate the sample container into the direction of flow and fill the sample container.
8. If rinsing the bottle, allow the sample container to fill at least one third of the container volume. Remove from the water and recap.
9. Shake the sample container gently and pour the water downstream of the sample collection point.
10. Complete the rinse procedure three times.
11. To fill the sample container, repeat steps 4 to 7 and then allow bottle to fill completely, turn the bottle upright and remove it from the water.
12. Replace the lid on the bottle.
13. Check that details on the sample container are correct.
14. Place the sample container in a cooler box (with ice or ice bricks) or refrigerator and chill. Double bag samples if ice is to be used.
15. Fill out the chain of custody form.

4.7.5. Manual sampling using water samplers

1. Label sample containers.
2. Collect in situ water quality measurements before collecting water samples for chemical analysis. Measurement of temperature, dissolved oxygen and conductivity can indicate the presence of stratification in the water, and may dictate where in the water column samples should be collected. See *Water quality*

- sampling using in situ water quality instruments document.
3. Inspect the internal surfaces of the water sampler, ensuring it is clean. If sampler has been washed prior to use, further rinsing of the water sampler is not necessary because this will occur as the device moves through the water body.
 4. Cock the water sampler and lower into a representative area of water and to the required depth.
 5. Trigger the water sampler as per the manufacture's instruction to fill, and remove the device from the water.
 6. To rinse the labelled sample container, pour water from the sampler into the labelled container until one third full. Replace the lid on the labelled sample container and shake gently. Remove the lid from the labelled sample container and pour the rinsate out downstream of the sample collection point.
 7. Rinse the sample container three times (if appropriate).
 8. Once the final rinse of the labelled sample container is complete, discard any remaining water from the water sampler away from the sample collection point.
 9. Refill by cocking the water sampler, lowering the sampler to the required depth and triggering the sampler.
 10. Remove the sampler from the water and fill the labelled sampler containers. Do this quickly to avoid sediment particles from settling to the bottom of the water sampler. Recap the sample containers.
 11. Complete a final check that details on the sample container are correct.
 12. Place the sample container in a cooler box (with ice or ice bricks) or refrigerator and chill. Double bag samples if using ice.
 13. Thoroughly rinse discrete depth sampler three times with high quality deionised water, allow to dry and store in a clean location prior to reuse at further sampling sites.
 14. Fill out the chain of custody form.

4.7.6. Filtering the sample

When filtering a sample there are a number of aspects to consider:

- Various types of filter membranes are available, and the choice of membrane depends upon the parameters being analysed. A cellulose acetate filter is commonly used to filter water samples that are being tested for metals, and polyethersulfone filters for nutrients. Contact the filter manufacturer for recommendations. Glass fibre pre-filters are not recommended for use when testing for trace or ultra-trace metals because of the contamination risk. The two most important aspects of filtration to consider are:
 - The working definition of a 'dissolved' sample (e.g. dissolved metals or dissolved organic carbon) is a sample that has been filtered through a 0.45µm membrane.
 - The filter is a common source of contamination. Make sure blanks are taken to test the quality of the filter. Further information is provided in Quality control for water and sediment sampling document.
 - Filtration cannot be undertaken on a sample that has preservative in it.
- If collecting a 'total' and a 'dissolved' sample it is recommended that the 'dissolved' sample be sub-sampled from the 'total' sample container, if preservative is not present in the container. This allows a cross check for the total and dissolved results. Further information is provided in the *Quality control for water and sediment sampling* document.
- If speciation studies are being undertaken, samples must be filtered immediately after collection. Nutrient samples should always be filtered immediately after collection if possible.
- Generally, samples should be filtered in the field. However, if trace or ultra-trace analysis is required, it may be more appropriate to filter samples in the evening (after return from the field) in a clean environment. The filtration area can be any room that is clean and dust free.

The procedure for filtering samples is:

1. Prepare a clean work area and ensure all equipment that you will require is unpacked and easily accessible in order to minimise the risk of contamination.
2. Label all sample containers.
3. Put on a new set of gloves immediately prior to the commencement of filtering.
4. To rinse the syringe, pull out the plunger and fill about a third full with sample water. Replace the plunger and gently shake to rinse the internal surfaces of the syringe and plunger. Repeat 3 times.
5. Remove the filter from the packaging and attach filter to syringe (see **Figure 12**).
6. If subsampling from a 'total' sample container:
 - 6.1. Shake the 'total' water sample gently to resuspend particulate matter.
 - 6.2. Place filter on the syringe and fill the syringe with sample water.
 - 6.3. Push a couple of millilitres of sample through the filter to rinse, and discard the rinsate away from the sample processing area.
 - 6.4. Push approximately 5mL of sample through the syringe and filter into the 'filtered' sample container.
 - 6.5. Replace the lid on the labelled sample container and shake gently to rinse all internal surfaces including the lid.
 - 6.6. Remove the lid and discard the rinsate away from the sample processing area.

- 6.7. Repeat the rinse of the 'filtered' sample container three times.
- 6.8. Shake the 'total' water sample gently and fill the syringe with sample water.
- 6.9. Remove the lid of the 'filtered' sample container and filter the required sample volume into the sample container.
7. If collecting directly from a waterway:
 - 7.1. Rinse the syringe three times in sample water.
 - 7.2. Fill the syringe with sample water and place filter on end of syringe.
 - 7.3. Push a couple of millilitres of sample through the filter to rinse.
 - 7.4. Push approximately 5mL of sample through the syringe and filter into the 'filtered' sample container.
 - 7.5. Replace the lid on the labelled sample container and shake gently to rinse all internal surfaces including the lid.
 - 7.6. Remove the lid and discard the rinsate away from the sample processing area.
 - 7.7. Repeat the rinse of the 'filtered' sample container three times.
 - 7.8. Fill the 'filtered' sample container with water pushed through the syringe and filter.
8. Recap the sample container.
9. Place the sample container in a cooler box (with ice or ice bricks) or refrigerator and chill.

The filtering process is illustrated in **Figure 12**.

Note: If water is turbid, filtering can take a long time.

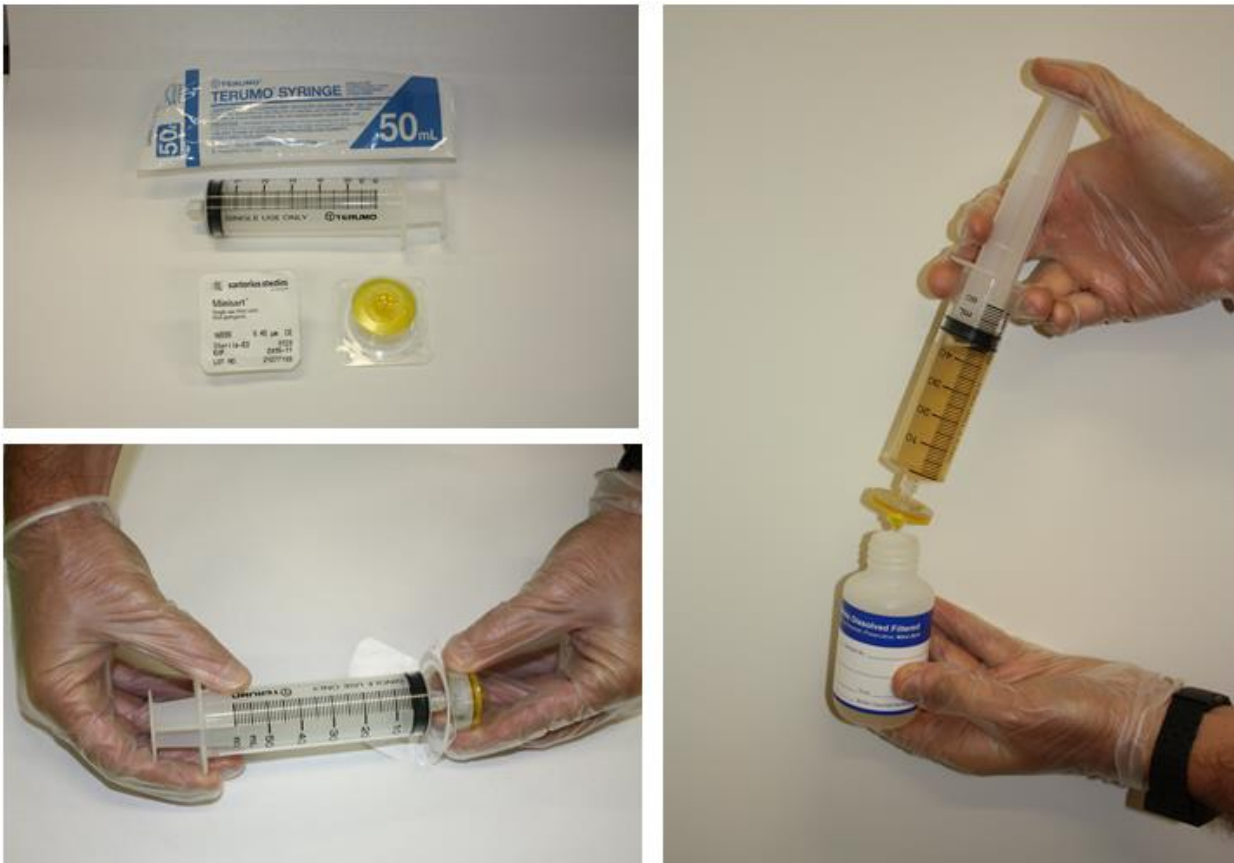


Figure 12: Filtering a water sample into a sample container

4.8. References and further reading

USEPA 1996, Method 1669, Sampling Ambient Water for Trace Metals at EPA Water Quality Criteria Levels, United States Environmental Protection Agency, Washington, viewed 30 August 2016, www3.epa.gov/caddis/pdf/Metals_Sampling_EPA_method_1669.pdf.

Appendix 1**Table 1: Equipment checklist**

Equipment	✓
Sample containers prepared by an accredited laboratory	
Non-powdered gloves	
Chain of custody forms, data sheets, notebook or field computer	
Water proof markers and pens	
Sampling pole with attachment to hold sample container	
Water sampler and associated equipment if appropriate	
Waders, gumboots or similar personal protective equipment (shore based sampling)	
Filters and syringes	

Background to water quality sampling using automated sampling equipment

Version: June 2018

5. Background to water quality sampling using automated sampling equipment

5.1. Purpose and scope

This document provides background information on water quality sampling using automated sampling equipment.

5.2. Associated documents

Physical and chemical assessment: Water quality sampling using automated sampling equipment

5.3. Introduction

Automated sampling devices are used to collect water samples when it is not possible to collect them manually. For example:

- when sampling flood waters where it is unsafe to collect a sample manually e.g. high river levels and high flows
- when sampling stormwater drains that can have flashy unpredictable flows
- where there is a requirement to sample at regular intervals throughout a 24 hour period. For example, to capture the rise or fall of a hydrograph for calculating loads.

In these situations an automated sampling device can be used to collect grab or composite samples. Types of automated sampling devices include refrigerated or non-refrigerated automatic pump samplers (auto-samplers) (**Figure 13a**) and rising stage samplers (**Figure 13b**). An understanding of the stream height at any given time is required to provide sample context. This may be via real time monitored river height data, or installed depth loggers which can be interrogated following flow events.

5.4. Automatic pump samplers (auto-samplers)

Auto-samplers (**Figure 13a**) are comprised of a number of bottles in a carousel or a single receptacle, a sample intake line that is fixed in place within the stream connected to a pump and a computer controlled data logger that requires programming.

The auto-sampler is programmed to be 'triggered' when a pre-entered set of conditions are met, for example a certain:

- stream height
- time of day
- change in temperature
- rate of rise or fall of the stream level
- turbidity reading.

Once triggered, the auto-samplers start sampling according to the program set.

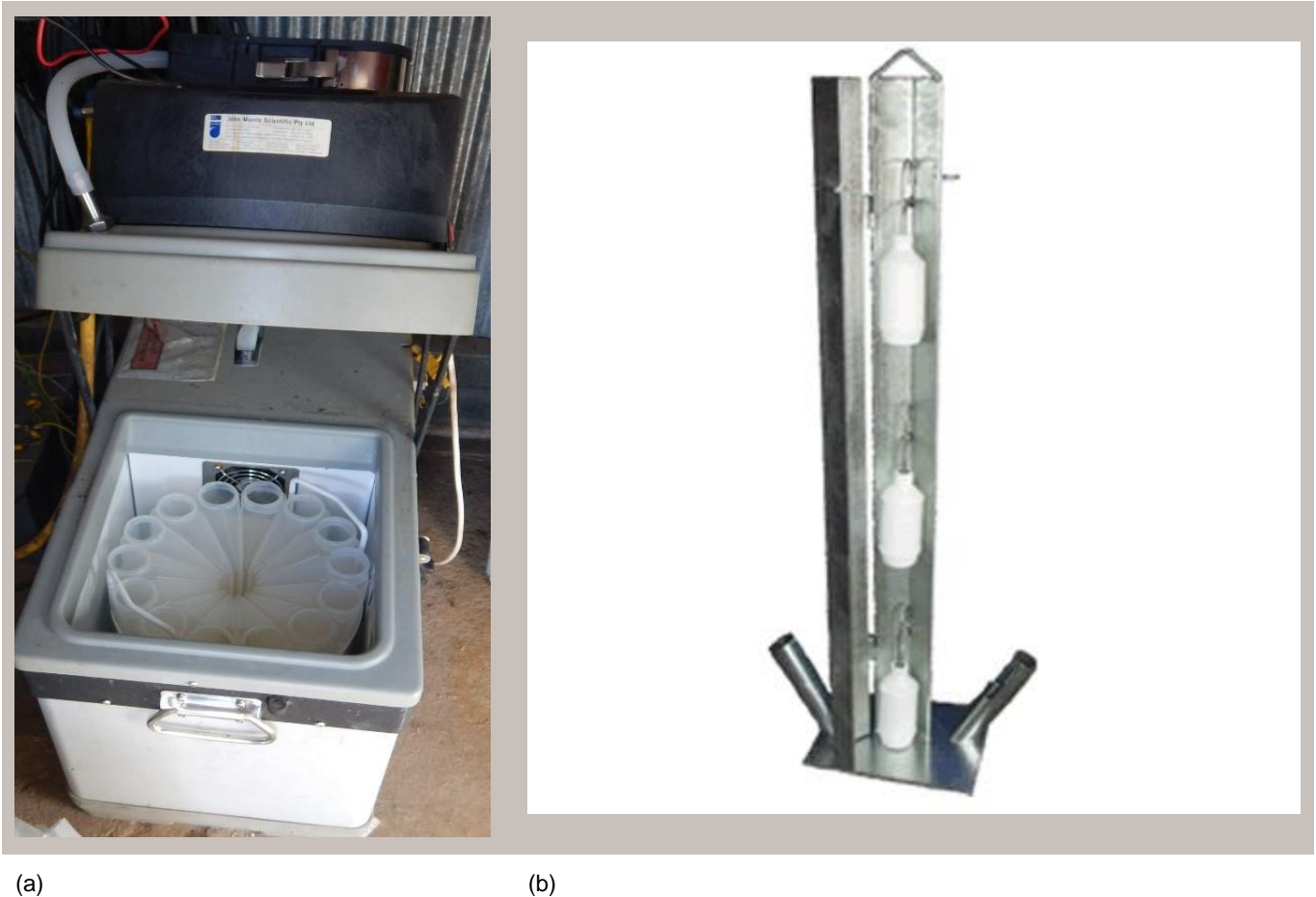


Figure 13: Automated sampling equipment: (a) refrigerated automatic pump sampler (Photo credit: DES) (b) rising stage sampler (Photo credit: Hyquest Solutions)

Installation of auto-samplers should be in accordance with the manufacturer's instructions. When using auto-samplers for collecting water samples, it is important to follow procedures for sample handling, sample holding times and sample containers for the analyte being measured.

When using auto-samplers, the possibility of exchange between the sample and the walls of the sampling lines (tubing or pipes) must be considered. The time of contact between the sample and the wall material is important and the automatic pump sampler must be programmed to flush out residues of previous samples before liquid is delivered into the sample container.

5.5. Rising stage samplers

Rising stage samplers (**Figure 13b**) are useful for collection of samples from flashy, intermittent streams. Rising stage samplers are not refrigerated, and so samples are exposed to ambient temperatures and often light. The use of rising stage samplers may not be suitable for the collection of samples for the analysis of a number of analytes, particularly where analytes rapidly degrade or transform (e.g. ammonium and nitrate).

Rising stage sampling units must be securely mounted, one above the other at known height locations relative to river height, with adequate support provided to prevent dislodging by large logs and other debris. Samplers should be erected and installed so that they are pointing in a downstream direction (ensure the inflow assembly of each bottle is facing upstream). Recommended locations to protect samplers are:

- on the inside of river bends (debris tend to be swept to the outside of bends)
- adjacent to large trees (providing partial protection)
- downstream of small shrubs and trees (provide partial protection).

Sampling units must be routinely inspected for evidence of insects in the intake and breathing tubes—a common reason for missed samples (and contamination). Sampling intakes and exhaust tubes should be replaced or thoroughly washed with laboratory supplied ultra-pure water at regular time intervals (monthly or sooner as required) to avoid blockages and contamination.

With rising stage samplers, water samples are taken as the river level rises and samples can only be retrieved after the river level has receded. As the water rises it reaches the intake tube (**Figure 14**) and begins to fill the bottle. Sampling ceases when the level of the water in the bottle reaches the inner end of the air exhaust, which then prevents circulation through the sampler. An air lock forms in the intake tube and prevents further water from entering the bottle. The rate of rise in the river height must not exceed the rate of filling of the rising stage sampler, otherwise only a partial sample will be collected before the air exhaust forms and air lock.

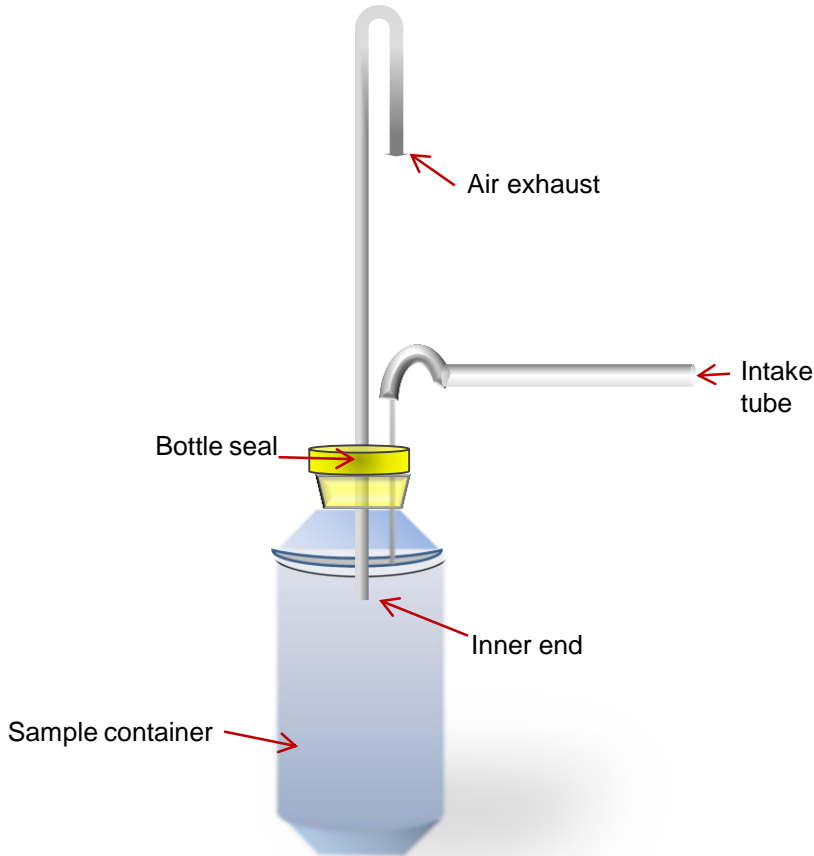


Figure 14: Schematic of the stage sampler bottle with intake and exhaust assembly

The date and time at which sampling waters have risen above the intake tube should be recorded as the sample date and time. This can be determined by analysing the hydrograph during or after the event. Samples should be collected as soon as accessible following an event to maintain sample integrity.

5.6. References and additional reading

AS/NZS 5667.1: 1998, Australian/New Zealand Standard Water quality—*Sampling Part 1: Guidance on the design of sampling programs, sampling techniques.*

Water quality sampling using automated sampling equipment

Version: February 2018

6. Water quality sampling using automated sampling equipment

6.1. Purpose and scope

This document describes the method for the correct use of automatic samplers when collecting water samples intended for laboratory analysis of water quality parameters. Automated sampling devices include refrigerated or non-refrigerated automatic pump samplers and rising stage samplers. This document describes the method for using both automated sampler types. There are limitations associated with automatic sampling (e.g. intermediate sampling containers, sampling from a fixed point, sample storage and preservation), which should be considered and documented.

6.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping including taking field photographs and videos*

Physical and chemical assessment: Background to water quality sampling using automated sampling equipment

6.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

6.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals for more information on requirements.

6.5. Skills, training and experience

Staff using the methods in this document must have previous training and experience in the use and calibration of automated sampling equipment. Ideally this training should be recorded in a central location. The operator should also be familiar with manufacturer's instructions.

6.6. Equipment

See Appendix 1 for example equipment checklist.

6.7. Procedure

Note: Auto-sampler bottles can be fitted with either polypropylene (**Figure 15**) or glass (**Figure 16**) sampler bottles depending on parameters being monitored.

6.7.1. Collecting samples using automatic pump samplers (auto-samplers)

1. On arrival at the field based automatic pump sampler (auto-sampler), halt the program.
2. Retrieve and record data from the auto-sampler e.g. sample details, storage temperature, and internal clock settings.
3. Ensure that the auto-sampler bottles are labelled in the order in which they were automatically filled to avoid errors when sampler bottles are removed.
4. Wearing clean powder-free gloves, place lids on each of the auto-sampler bottles while still in the carousel. Remove individual auto-sampler bottles in sequence from the carousel, then follow steps 5-7 for each bottle.
5. Depending on the parameter being monitored, laboratory supplied sample bottles may need rinsing before the water sample is decanted. If so, shake the auto sampler bottle vigorously to resuspend settled material, decant a small volume of sample from the auto-sampler bottle into a laboratory supplied sample bottle, recap the laboratory supplied bottle and shake to rinse. Discard the rinse water

Note: Do not rinse bottles that contain preservative.

6. Repeat Step 5 twice more.
7. To collect the sample, recap the auto-sampler bottles, shake to resuspend settled material and decant into the rinsed laboratory supplied bottle. Replace the lid on the laboratory supplied sample bottle and keep refrigerated or on ice bricks until the sample can be stored/preserved appropriately before dispatch to the analytical laboratory.
8. If enough sample remains in an auto-sampler bottle, other samples can be collected for additional analyses. Record what samples are collected from each auto-sampler bottle.
9. If you intend to change the volume of sample to be collected by the auto-sampler you will need to refer to the manufacturer's operating procedures. Follow the manufacturer's operating procedures and calibrate the auto-sampler using a graduated measuring cylinder.
10. Fill the auto-sampler with clean bottles and store clean lids appropriately to mitigate contamination.
11. Restart the auto-sampler program ensuring the distributor arm (for sample delivery) will begin filling at bottle 1.
12. Dispatch water samples and necessary paperwork to the analytical laboratory for analysis within specified holding times. Ensure samples are packaged and transported within a timeframe appropriate to maintain storage conditions (e.g. filtered nutrients need to be kept chilled or preferably frozen to maintain parameter stability).
13. Clean used auto-sampler bottles. Use a soft bristle brush to dislodge residual particulate matter and allow bottles to soak in phosphate free detergent solution for three hours. Scrub internal and external surfaces of bottles and lids. Rinse bottles and lids with deionised water three times and allow drying in a clean location. If pump sampler bottles are glass, conduct a further final rinse using an appropriate solvent for the analyte of interest.
14. Store clean, dry automatic pump sampler bottles appropriately to minimise contamination.



Figure 15: Polypropylene pump sampler bottles



Figure 16: Glass pump sampler bottles

6.7.2. Collecting samples using rising stage samplers

Note: Rising stage samplers should be loaded with suitably cleaned bottles to minimise contamination. It is possible to have the rising stage sampler fittings modified to accept bottles supplied by a laboratory.

1. Bottles must be labelled with a unique identifier.
2. If the rising stage sample bottle is used as an intermediate container (to fill other sample bottles) follow steps 5-8 in Section 6.7.1. Record what samples are collected from each bottle.

3. If the rising stage sampler has been modified to accept laboratory bottles, simply remove the inflow assembly and put rinsed laboratory supplied lid on bottle. Make a record of each bottle collected from the site.
4. When removing the stage sampler bottle, replace the inflow assembly (**Figure 17**) of the stage sampler bottle with a clean lid.
5. When cleaning the stage sampler bottles, ensure all internal surfaces of the bottles, lids, hoses and pipes are scrubbed clean after soaking in a phosphate free detergent solution for three hours. Rinse the bottles and inflow assemblies with distilled water three times. Particular care must be taken to thoroughly rinse the inflow hoses and pipes.



Figure 17: Stage sampler bottle with inflow assembly

6.8. References and additional reading

AS/NZS 5667.1:1998, Water quality – Sampling: Guidance on the design of sampling programs, sampling techniques and the preservation and handling of samples, Standards Australia.

Appendix 1**Table 1: Equipment checklist**

Equipment	✓
Clean, powder-free gloves	
Data sheets, notebook or field computer	
Waterproof marking pen	
Vehicle mounted fridge or Eskys containing frozen ice bricks	
Sample bottles of suitable composition and pre-treated for the analysis required	
Clean replacement automatic sampler bottles	
Soft bristled brush	
Phosphate free detergent and associated safety data sheet (e.g. Decon 90)	
Solvent and associated safety data sheet (e.g. methanol GR for analysis)	
Laboratory supplied deionised water	
Graduated measuring cylinder (minimum 2L volume)	
Manufacturer's instruction manual for automatic pump sampler	

Background to the use of passive sampling methods for trace contaminant detection

Version: February 2018

7. Background to the use of passive sampling methods for trace contaminant detection

7.1. Purpose and scope

This document describes passive sampling concepts for monitoring trace concentrations of contaminants. Passive sampling devices (PSD) are used to:

- detect contaminants that may be present in concentrations below the limit of detection that a laboratory can reach when testing a water sample. Trace levels of contaminants are often concentrated to detectable levels by passive samplers placed in water for a controlled exposure period.
- obtain a time-weighted average concentration over a deployment period, which can vary between several days and several weeks for different passive sampler types and for different analytes.

7.2. Associated documents

Manufacturer's instructions for use of passive sampling devices (PSD).

7.3. Introduction

In principle the use of a PSD involves the deployment of a chemical-absorbing or -adsorbing material (accumulating material) in the water column (or sediments). After a period of exposure, the sorbent material is retrieved and the accumulated chemicals are extracted/eluted from the sorbent phase and analysed. The basic components of a PSD are:

- an accumulating medium for the analyte/s of interest
- a membrane to control the rate of uptake (in the case of samplers designed for polar chemicals) that can also act as a protective layer
- a mounting structure to contain and protect the other components but at the same time expose them to the environment being sampled.

Some PSDs will also have a transport medium between the membrane and the accumulating medium to ensure diffusion-controlled and consistent rates of uptake. This is more common in PSDs for inorganic contaminants, which tend to be present at higher concentrations than many organic contaminants.

Passive sampling devices can be deployed in the field in a variety of situations—hung from floats, suspended from jetties, fastened to stakes inserted in a stream bottom, embedded in sediment, or anchored to the bottom but held up into the water column by a float.

The normal requirements of Quality Assurance and Quality Control apply when sampling with passive sampling devices. These include the use of trip blanks, replicate samplers, and the use of personal protective equipment such as gloves to avoid contamination of the sampler or its housing during handling. It is also important to assess the surrounding environment for contamination as some passive samplers can adsorb compounds from the atmosphere. Further information can be obtained from the BSI Standards Publication detailing - Guidance on passive sampling in surface waters (BSI 2011).

The main source of variability when using a PSD is the extent to which water flow conditions around the sampler affect the degree of sorption or desorption. Several approaches are used to determine the effect of flow on the chemical accumulation to increase confidence in the results. These include:

- The use of performance reference compounds for organic pollutants, which are introduced into the accumulating medium of sampler devices before deployment. Performance reference compounds are selected to have similar diffusion and accumulation properties as the chemicals to be sampled (i.e. similar octanol/water partition coefficients). The rate at which these compounds are lost from the sampling device are assumed to be proportional to the rate of uptake of organic analytes.
- Water flow information can be obtained from rate of dissolution of a slightly soluble compound or material (e.g. plaster of Paris) deployed alongside the PSDs.
- PSDs with a diffusional medium can deploy samplers with several different thicknesses in order to obtain an

average diffusion boundary layer thickness, which also relates to flow.

7.4. Passive sampling for organic substances

Passive sampler devices for detecting organic chemicals in water have evolved over many years, and various devices and methods have been employed. Most of these methods fall into two categories: those that use an organic solvent as the sorbent phase, and those that use a solid sorbent phase.

7.4.1. Semi-permeable membrane device

The most widely used passive sampler design for non-polar chemicals in water is the semi-permeable membrane device (SPMD) (**Figure 18**). It consists of a length of sealed polyethylene tubing (the membrane) containing a small volume of triolein (the sorbent phase) woven around a stainless steel frame. The device is then inserted into a perforated stainless steel shroud for protection from mechanical damage during deployment. This design allows a very high surface area to maximise rates of accumulation.

An alternative to the triolein-based SPMD described above is to use a strip of silicon rubber such as polydimethylsiloxane (PDMS) placed inside the deployment shroud as the absorbent material.

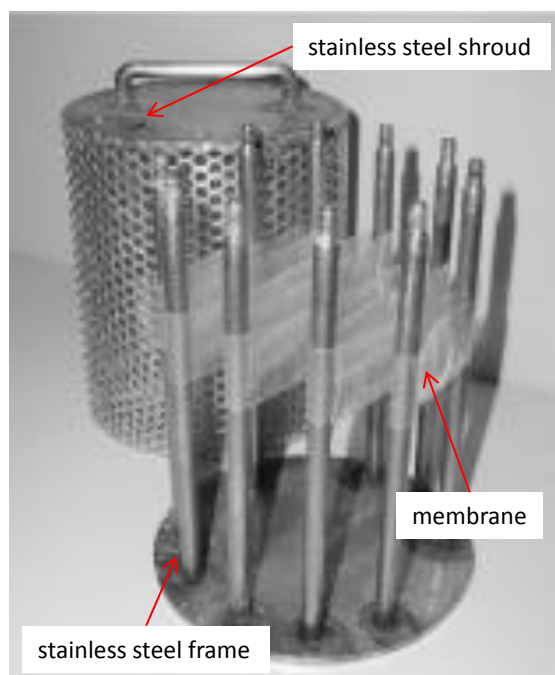


Figure 18: Housing and components of an SPMD passive sampler. The protective shroud covers the absorbent strip during deployment

7.4.2. Chemcatcher®

This device is a very robust PSD that employs solid phase extraction disks (e.g. Empore™ disks by 3M™) such as bonded silica (e.g. C18) or poly(styrenedivinylbenzene) copolymer (e.g. SDB-RPS) as the sorbent material, combined in most cases with a polyethersulfone (PES) membrane that allows diffusion of polar chemicals. One of these devices based on the Empore disk (**Figure 19**).

The sampler consists of three interlocking sections (2, 3, 9) manufactured from polytetrafluoroethylene (PTFE) that screw together during deployment to form water-tight seals (4, 10). Integral to the device is a 50mm rigid PTFE disk (7) designed to support both the sorbent material (Empore) (5) and the diffusion-limiting membrane (6). On the reverse is a lug (1) for attaching the device during deployment. The outer surface of the diffusion-limiting membrane is protected from mechanical damage during deployment by a mesh (8) of either stainless steel for organic analytes or nylon for inorganic analytes. This mesh is held in place during deployment by a removable PTFE ring (9). A PTFE screw cap (11) replaces the ring (9) during transport to and from the deployment site.

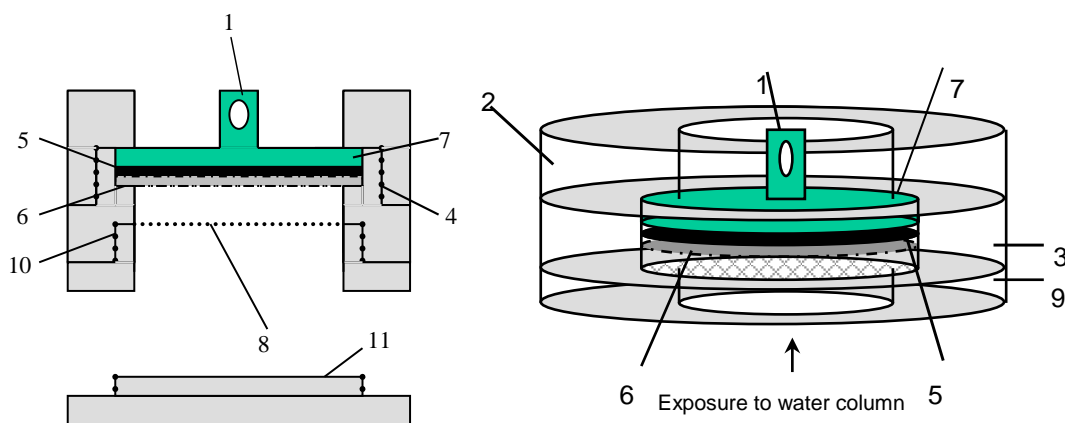


Figure 19: Housing and components of a Chemcatcher® passive sampling device

7.5. Passive sampling for inorganic substances

The diffusive gradient in a thin film (DGT) device employs a binding layer, which is typically a polyacrylamide hydrogel that is cast containing an accumulating material (such as a chelating resin or metal oxide adsorbent). The binding layer or gel is overlaid by a transportation medium (diffusive layer or gel) to maintain a concentration gradient over a known thickness and a filter to exclude particulates (**Figure 20**). Numerous binding layers have been developed to measure a range of metals (cations and oxyanions), dissolved inorganic nutrients (phosphate, nitrate and ammonium), sulphide and radioisotopes. DGT devices can also be deployed with several different diffusive gel thicknesses which allows accurate measurement under low flow conditions.

An important advantage of using DGT to measure metals in saline or marine waters is that the binding layers do not accumulate the major ions that often cause interferences in the analysis of metals. The devices sample satisfactorily over a range of pH with the range limits varying between metals – these have been well-defined for most common metal analytes. The DGT device is deployed for much shorter time periods than organic PSDs as inorganic analytes tend to be present at higher concentrations and to ensure linear uptake.

A similar PSD, known as DET (diffusive equilibrium in thin films), which does not contain a binding layer, can be deployed in sediment (as can DGT) for solutes for which there is no suitable binding layer. The DET comprises a single relatively thick sheet of gel (typically 0.8mm) supported in a holder with a membrane. Solutes in the surrounding water diffuse into the gel until concentrations equilibrate.

The University of Lancaster (UK) holds a worldwide patent on both DGT and DET devices but they are both available commercially, either pre-assembled or in kit form (gel disks and strips for local assembly). For details of supply visit the DGT for measurements in waters, soils and sediments website at www.dgtresearch.com.

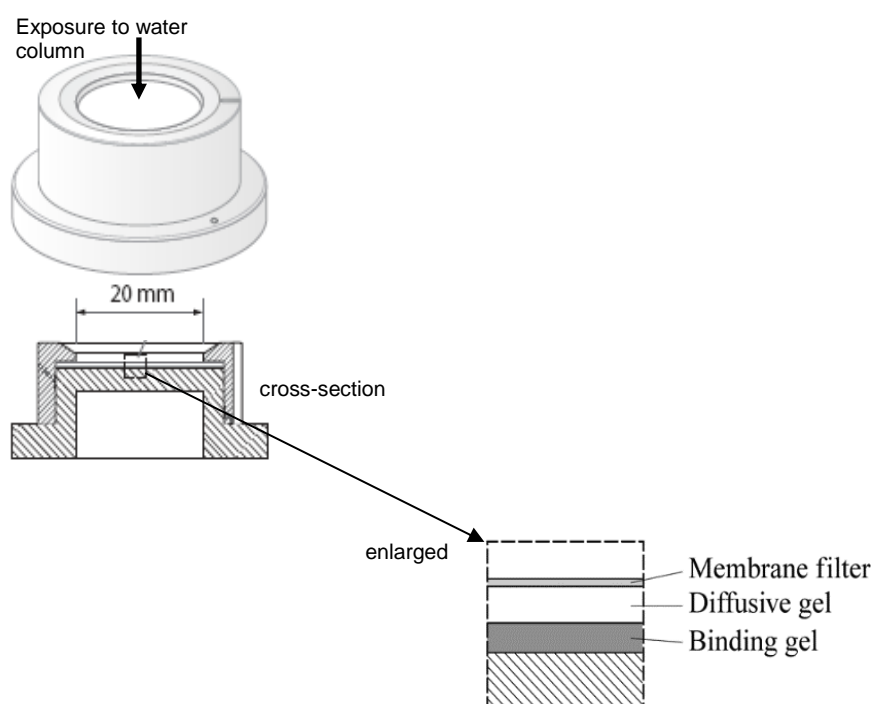


Figure 20: Housing and components of a DGT passive sampling device

7.6. References and additional reading

BSI 2011, Water Quality – Sampling. Part 23: Guidance on passive sampling in surface waters (ISO 5667-23:2011), BSI Standards Publication.

Background to event monitoring

Version: February 2018

8. Background to event monitoring

8.1. Purpose and scope

This document provides some background information on event monitoring.

8.2. Associated documents

Sampling design and preparation:

- *Sampling scope and design*
- *Preparation for sampling*

Physical and chemical assessment:

- *Manual collection of surface water samples (including field filtration)*
- *Background to water quality sampling using automated sampling equipment*
- *Water quality sampling using automated sampling equipment*
- *Collection and preservation of sediment*

8.3. Introduction

Event monitoring is the collection of water quality and quantity information during periods of increased waterway⁶ discharge⁷, with a primary objective of estimating loads of contaminants (e.g. sediments and nutrients) that are transported during events. A discharge event can be caused by various factors (e.g. rainfall runoff⁸, groundwater discharges to surface waters, water storage releases) and may mobilise and transport increased amounts of contaminants. Water quality and quantity monitoring during these events is often referred to simply as event monitoring. Contaminant loads are known to vary according to a range of factors including hydrological flow (Brodie et al. 2010, Peljo et al. 2013), spatial distribution of rainfall and rainfall intensity, size of the catchment, topography (Young et al. 1996), antecedent soil moisture conditions (McDowell and Sharpley 2002), catchment land use (Packett et al. 2009), geology and soil type (Caitcheon et al. 2001), and the proportion and type of ground cover (Bartley et al. 2006).

Discharge events occurring at the beginning of a wet season may result in high fluxes of contaminants. This is known as a first flush effect, which occurs as a result of the transport of material accumulated during the dry season. As a result of low rainfall, ground cover vegetation may be reduced by the end of the dry season. Rainfall onto bare soils may mobilise soil particles and contaminants held in the soils, leading to infiltration and runoff during the first rainfall event of the season. Later in the wet season, ground cover is expected to be increased. Ground cover vegetation slows the rate of surface water runoff allowing more infiltration to occur. This reduces the ability of rainfall to mobilise contaminants attached to surface soils. Variability between events may also be compounded by land management practices, including stocking rates, harvesting, fertiliser application and seasonal land preparation regimes.

8.4. Why monitor events?

Data obtained from an event monitoring program may be used to:

- monitor catchment loads and link these to management actions to reduce contaminant loads (e.g. Water Sensitive Urban Design, riparian rehabilitation, nutrient offsets and on ground works)

⁶ Waterways for event monitoring purposes are any rivers, streams or creeks – and their tributaries – including the water, channel and riparian zone; estuaries and wetlands.

⁷ Discharge (streamflow) is the volume of water that flows past a cross-section of the stream over a unit of time.

⁸ Rainfall runoff is produced when rainfall intensity exceeds the infiltration capacity of the soil causing water to flow overland and into waterways. Runoff can be influenced by various characteristics (e.g. rainfall (intensity, duration, distribution), soil type, vegetation, slope and catchment size) and result from weather events ranging from small, high intensity localised storms and flash flooding to prolonged rainfall associated with tropical lows and cyclones.

- aid in understanding catchment processes (e.g. water quality, nutrient cycling, erosion, sediment/nutrient transport and landuse change)
- identify contaminant sources (e.g. point and diffuse sources)
- better characterise diffuse contaminant concentrations during events from different sources and under a range of flow conditions
- provide data on contaminant generation from major land uses, land use change or intervention in the catchment upstream of the monitoring sites
- calculate contaminant loads for parameters of concern
- provide data for license, permit, environmental authority requirements
- adjust and calibrate values used in catchment models and provide locally specific data for parameters of concern.

8.5. What to monitor

To identify the contaminants you wish to monitor, region specific factors affecting contaminant loads (listed previously) should be characterised. Water quality data, focused on contaminants of interest, needs to be collected through intensive monitoring over a variety of events that differ in size and timing – e.g. largest and smallest events, first event of the season – and over a period that enables the capture of seasonal variability. Discrete and continuous sampling during events also needs to cover the different stages of the waterway discharge hydrograph – i.e. baseflow, the rising stage of discharge and the falling stage of discharge – and capture the concentration variations of the contaminant of interest. When event monitoring, the quantity of water flowing down the waterway also needs to be measured as an important component of loads calculations, and to aid interpretation of contaminant concentration data. Data from established gauging stations are available from the Queensland Governments Water Monitoring Information Portal (WMIP) (<https://water-monitoring.information.qld.gov.au/>) or the Bureau of Meteorology (<http://www.bom.gov.au/water/>).

8.5.1. Event water quality sampling

Event monitoring aims to collect data from as many flow events as possible in order to calculate loads of contaminants. A large proportion of contaminant loads are generated during the wet season. Although large flood events produce high loads, in some circumstances, significantly higher contaminant concentration peaks can occur in small discharge events (**Figure 21**), particularly the first discharge event of the season (first flush). Sampling should be done at a frequency to maximise the accuracy of calculating contaminant loads over the hydrograph, noting that the peak of contaminant concentrations may not necessarily correlate with the peak of discharge.

When monitoring a new site, the relationship between the rise and fall in waterway height and the associated change in contaminant concentrations will not be known. Intensive sampling should therefore be undertaken in the early stages of sampling to determine the characteristics of the site. Up to 20 samples over an event may be required to gain this knowledge. Once a good understanding is established, it may be possible to carry out future monitoring at the site with fewer samples while still ensuring accuracy in estimating total loads. For example, a minimum of six samples, with most samples collected during the falling stage (for the Great Barrier Reef Region), may be enough coverage of the event to produce accurate total load estimations (Thomson et al. 2012). This would, however, be catchment specific and applicable to simple hydrographs only.

Installation of refrigerated auto-samplers that can be programmed to trigger sampling at a particular time or river height, are extremely useful in ensuring hydrographs are well sampled at a given site (see Background to water quality sampling using automated sampling equipment document).

Water quality samples should be delivered to a National Association of Testing Authorities (NATA) accredited laboratory for analysis within the prescribed holding times. Holding times will vary depending upon the analyses to be performed. See Choosing a laboratory and analytical method, holding times and preservation for further information.

Results obtained from the analytical laboratory will be assessed for quality control/quality assurance prior to data analysis and interpretation.

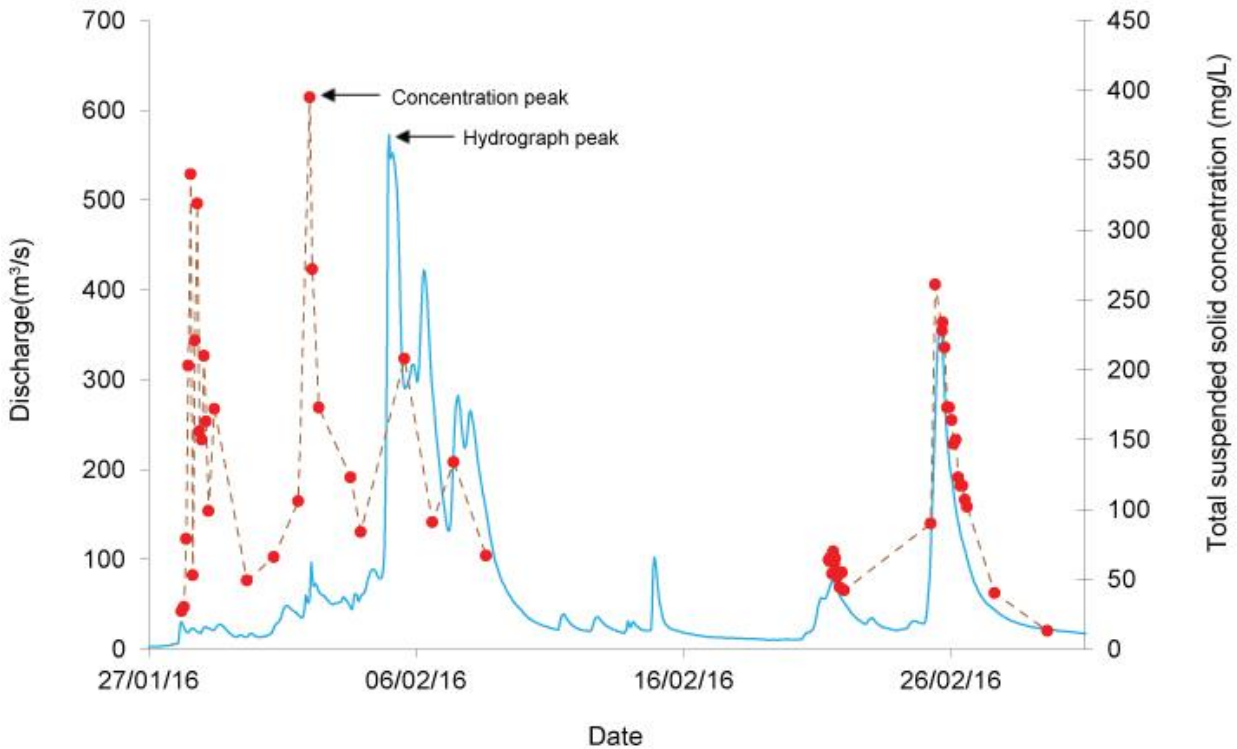


Figure 21: Example hydrograph showing the concentration of total suspended solids (TSS) (dotted line) against discharge (blue line). Red dot indicates the date samples were collected.

8.5.2. Baseflow water quality sampling

Also known as Dry Weather Concentration (DWC) sampling or ambient sampling, baseflow sampling differs from event sampling in that sampling is carried out when the flow is predominantly influenced by groundwater discharge and the overland flow component is small (i.e. no flow derived from rainfall runoff). Baseflow sampling may direct the effects of water and associated contaminants filtering into streams via groundwater and effects of riparian vegetation (e.g. leaf litter) on water quality. Light levels, temperature changes, groundwater recharge rates and instream processes can also affect water in the receiving waterway.

Sampling streams during baseflow should be undertaken with adequate lag time following an event to ensure the samples are solely baseflow water and not representative of the tail of an event. For baseflow sampling, a sampling frequency of once per month is usually adequate, bearing in mind this frequency is program specific and can be adjusted according to the programs objectives.

Total contaminant loads are calculated using the results of a combination of event and baseflow sampling over the defined period. In some systems, baseflow can contribute significantly to annual loads, so it is important to include baseflow water quality sampling when planning a monitoring and sampling schedule.

8.5.3. Hydrological measurements

Measurements of the stream's discharge volume are usually obtained by stream gauging techniques. Gauging is the process of measuring the volume of water flowing through and past a cross-section of a stream, within a defined time period. Refer to AS/NZS 3778.3.3:2001 for further details. Hydrological measurements and applications

Due to the impracticality of directly measuring waterway volume during high discharge events (due to workplace health and safety issues, access restriction or other hindrances), measurements may be carried out indirectly. This can be achieved by regularly recording the stream water level (stage height), using an automatic logger. This height is then related to discharge by applying an appropriate rating curve. It should be noted that this theoretical rating curve should still be validated by direct discharge measurements (gaugings) in order to reduce any error associated with the resulting discharge data.

8.5.3.1. Stream height

Continuous measurement of stream height should be recorded at sampling sites or at gauging stations close to the sampling site. These measurements are collected using an in-stream pressure transducer or gas bubbler system connected to a data logger. The pressure transducers should be selected with a designated calibrated range to

cover the expected stream heights; this will ensure an accurate recording of the hydrograph. The pressure transducers should have an accuracy of +/-30mm (water height). Pressure transducers used for stream height recording should be calibrated according to the manufacturer's instructions to ensure accuracy.

8.5.3.2. Discharge

Gaugings taken at sampling sites or hydrographic gauging stations (wherever pressure transducers are installed) are used to validate the rating curves that help to convert continuous height records into time series stream discharge data. All discharge measurements should be carried out and calculated in accordance with Australian and international standards. Gaugings during events should only be attempted where and when it is safe to do so in accordance with set workplace health and safety procedures. Gaugings should be performed only by trained hydrographic staff.

8.5.3.3. Rating curves

The development and application of an accurate discharge rating curve for a stream gauging station is critical to the production of reliable stream discharge data. Rating curves should be developed according to Australian and international standards and be properly recorded and preserved as a permanent record.

Software programs are available that can be used to develop theoretical and/or empirical rating curves using physical waterway parameters such as area, slope and roughness. These rating curves then need to be continually validated by gaugings throughout the range of the hydrograph. Rating curves are then applied to the height data using the software program to produce discharge data. Physical waterway measurements such as stream cross section and stream slope should be carried out in accordance with Australian and international standards.

Waterway discharge or water quantity measurements, including stream height, change over time, and the physical measurement of width, depth and velocity of the stream should be collected and used in accordance with Australian and international standards (AS/NZS 3778.3.3:2001 and ISO 1070:1992). These methods should be considered when:

- designing waterway gauging stations or monitoring sites
- maintaining and calibrating equipment for waterway discharge measurements
- using direct methods for discharge measurements (e.g. gaugings, acoustic doppler channel profiler (ADCP))
- using indirect methods for discharge measurements (e.g. area velocity method, flow factoring)
- developing and applying rating curves.

The quality and accuracy of the data should be determined, followed by the application of appropriate quality codes to the data.

8.5.4. Event sample/baseflow sample/field measurement metadata

All samples and measurements collected for an event monitoring program should be collected in association with standardised metadata. Guidance as to the metadata that should be collected in association with water quality parameters can be found in the National Industry Guideline for water quality metadata (Bureau of Meteorology 2016).

Note: It is essential that site, date and the exact time of collection are recorded in order to assess data against the hydrograph.

8.6. Data analysis and interpretation

Empirically-derived load estimates must be based on robust, accurate and repeatable loads determination methods (Marsh 2011). The accuracy of the resulting load estimate depends on how well the concentration data are able to characterise what is, in reality, continuously varying contaminant concentrations in the waterway. The accuracy of this characterisation depends on the number and timing of the collection of samples, the variation of the actual concentrations and the mathematics of how the flow and concentration estimates are combined. Where loads are required for all events and for each of the parameters typically measured (e.g. total suspended solids and nutrients), it is often not practical to undertake such detailed assessment to account for the high spatial, temporal, hydrological, geological and meteorological variability, and as such, there is no national, state or regional guidance on which method should be used to calculate loads.

Data analysis methods vary according to program objectives. Standardised methods are required that are specifically tailored to answer the original program objectives. This will ensure that any interpretation of event and baseflow data is meaningful. For the objectives of an event monitoring program, data analysis methods could include but are not restricted to:

- concentration and flow relationships (regression analysis)

- load calculations:
 - annual loads
 - event loads
 - daily loads
- land use yields
- Event Mean Concentrations (EMC)
- Site Mean Concentrations (SMC).
- Standardised analysis procedures and methods should be collaboratively developed and peer reviewed to ensure suitability for addressing original objectives.

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Background information on Acoustic Doppler Current Profilers (ADCPs)

Version: February 2018

9. Background information on Acoustic Doppler Current Profilers (ADCPs)

9.1. Purpose and scope

This document provides some background information on Acoustic Doppler Current Profilers (ADCPs) and their use for creating flow and total suspended sediment concentration profiles. ADCPs are instruments commonly used to measure the velocity of flow in rivers and creeks. Detailed instructions on the installation and use of ADCPs are available in National Industry Guidelines for the Application of Acoustic Doppler Current Profilers to Measure Discharge in Open Channels (WISBF GL 100.08-2013) and Application of in-situ Point Acoustic Doppler Velocity Meters for Determining Velocity in Open Channels (WISBF GL 100.09-2013)⁹. These standards should be followed.

9.2. Using ADCPs for continuous flow measurement at a fixed point

ADCPs are instruments commonly used in the collection of stream velocity data. They function by sending acoustic pulses from their transducer faces into the water column, where they are reflected back by particles and the exact time of their return to the transducer face is measured. The time of the returning signal is affected by the doppler effect of the moving particles in the water. The time differences measured in the returned signal are used to infer the velocity of the water in which the particles are travelling.

There are two types of fixed ADCP units (also known as acoustic doppler velocity meters or ADVM) that may be permanently mounted:

1. A horizontal ADCP (H-ADCP), which is mounted on the bank of a waterway. It is used to measure the velocity of a fixed horizontal slice of a cross section of the river (**Figure 22** and **Figure 23**).
2. A bottom-mounted ADCP (B-ADCP), which is mounted on the bed of a waterway. This is used to measure the velocity of a fixed vertical slice of the river (**Figure 24**).



Figure 22: Example of a horizontal Acoustic Doppler Current Profiler

⁹ <http://www.bom.gov.au/water/standards/niGuidelinesHyd.shtml>

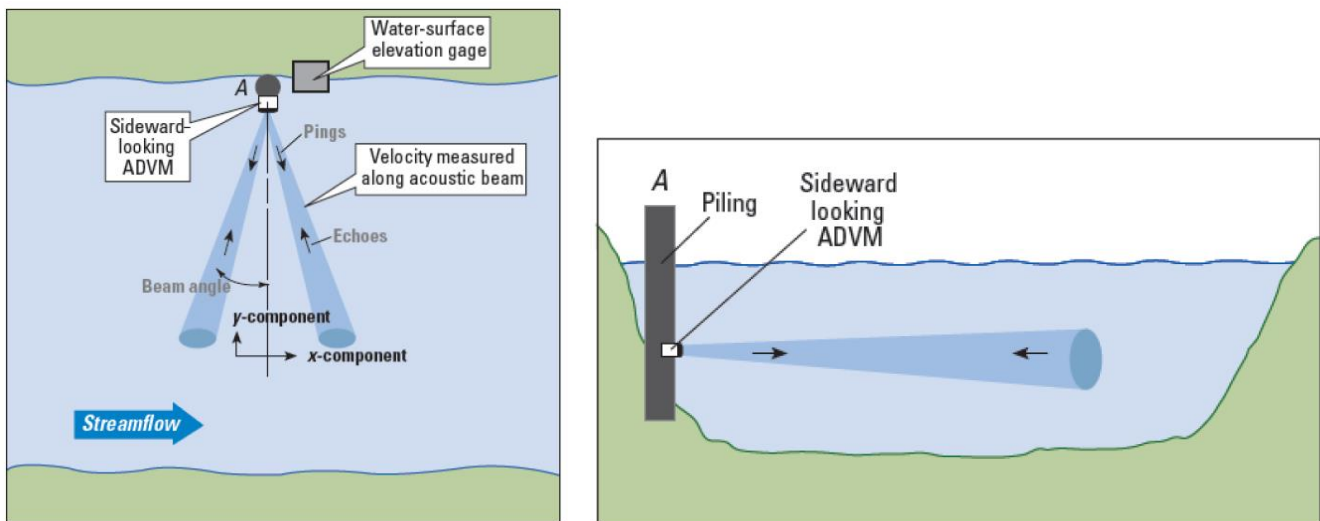


Figure 23: Bird's eye view (left) and channel view (right) - illustration of a horizontally mounted ADCP installation (Source: Ruhl and Simpson 2005)

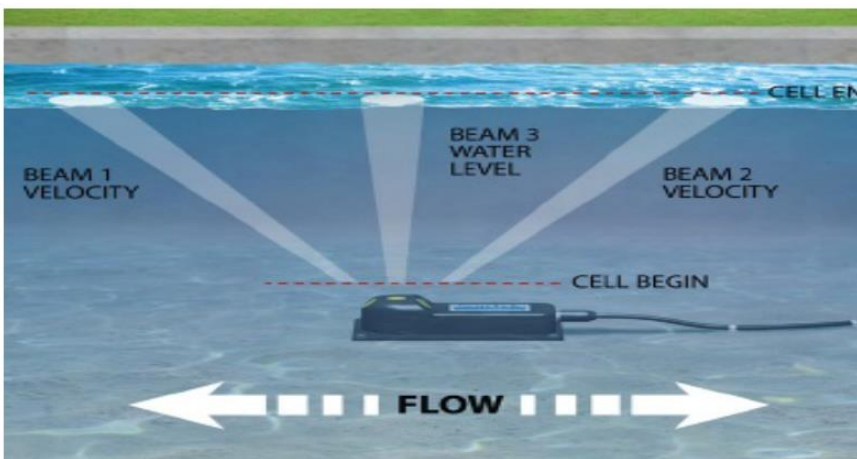


Figure 24: Illustration of a typical bed-mounted ADCP installation (Source: Sontek/YSI Argonaut product brochure, 2009. www.sontek.com/pdf/brochures/Argonaut-SW-2009-06-23.pdf)

The distance across the channel for which a H-ADCP can be used to make velocity measurements is dependent on the unit frequency and river depth. The choice of unit should always be one that can measure velocities across the entire channel width where possible. Bottom mounted ADCPs must be installed in the section of the channel that represents the mean velocity. During different flow events this location may change.

Because ADCPs are generally installed in a fixed location, changes in the river stage¹⁰ will also change the spatial variation of the velocity structure. It is therefore necessary to correct the measured H-ADCP and B-ADCP velocities for these changes to ensure reported river discharges are accurate. This is most commonly achieved by measuring the flow of the river under a range of flow conditions using a boat mounted ADCP (**Figure 25**) at the same time that the fixed ADCP is collecting data. From this, an index-velocity relationship can be established and in conjunction with the site stage-area relationship, can be used to correct the stream flow data measured and reported by the fixed ADCP.

An index-velocity relationship is specific to an installation height. If the H-ADCP or B-ADCP is removed and replaced at an alternative height or location within the same channel then the index-velocity equation is no longer valid and a new relationship is required.

The measurement of the channel's mean velocity at different stages using a boat mounted ADCP (**Figure 25**) is beyond the scope of this document, but should be carried out in accordance with the National Industry Guidelines

¹⁰ Stage is the water level above some arbitrary point, usually with the zero height being near the river bed.

(Section 1).



Figure 25: A boat-mounted ADCP prepared to collect velocity data

9.3. Using ADCPs to estimate cross sectional suspended sediment concentrations

In addition to the flow velocity data calculated by the ADCP, data related to the 'noise' of the returned acoustic signal is typically recorded by ADCPs. This data is commonly called 'backscatter'. If water quality grab samples are collected in conjunction with an ADCP measurement, it may be possible to correlate the total suspended solids (TSS) contained in the water quality samples, and the backscatter signal of the particles in the water. If land use upstream of the fixed ADCP is reasonably stable, and sufficient suspended sediment data is collected across a range of flow conditions (ambient to flood events), it is possible to generate estimates of continuous TSS concentrations at a site using either a permanent horizontal or bottom mounted ADCP. TSS samples can be collected using an isokinetic sampling device (for further guidance see USGS 2006).

To calibrate the ADCP backscatter data with TSS concentrations, it is necessary to collect grab samples at the same location that the ADCP is recording backscatter data. Calibration grab samples must be collected at times of differing levels of backscatter intensity to ensure robust inferred TSS concentrations calculations. **Figure 26** demonstrates the positions within a channel profile where grab samples need to be collected to provide an accurate calibration of a boat mounted ADCPs back scatter.

Grab samples need to also confirm that the increase in backscatter is due to increased TSS concentrations and not increased particle size.

Note: The installer/operator should be aware that there is an area directly in front of the ADCP transducers known as the 'blanking distance'. The blanking distance is a zone where data is disregarded due to the potential of signal interference. The size of the blanking distance depends on the instrument configuration, frequency and waterway conditions.

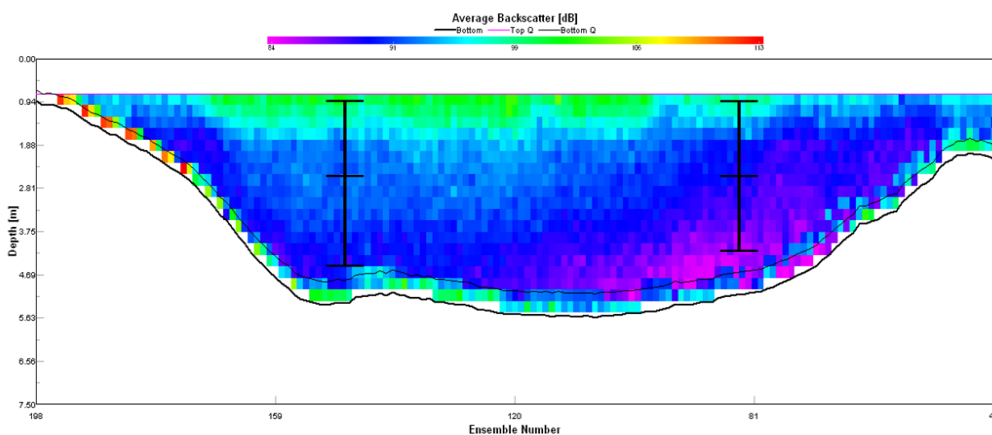


Figure 26: A channel profile showing the spatial variation in the measured backscatter intensity using a boat mounted ADCP. The black bars represent the three locations in each vertical where grab samples must be collected to accurately calibrate backscatter intensity with TSS concentrations (Source: Mark Randall, Department of Natural Resources and Mines, QLD)

9.4. References and additional reading

National Industry Guidelines for hydrometric monitoring. Part 8: Application of Acoustic Doppler Current Profilers to Measure Discharge in Open Channels (WISBF GL 100.08-2013). Available from:
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Chlorophyll a sample collection methods

Version: February 2018

10. Chlorophyll a sample collection methods

10.1. Purpose and scope

This document describes the procedure for field collection of surface water samples for subsequent laboratory extraction and analysis for chlorophyll a. The aim is to collect water samples from which a known volume of water is filtered under suction onto filter paper for subsequent analysis of chlorophyll a.

10.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping including taking field photographs and videos*

Physical and chemical assessment: Manual collection of surface water samples (including field filtration)

Biological assessment: Sampling freshwater and marine microalgae and harmful algal blooms (HABs)

10.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

10.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

10.5. Skills, training and experience

No skills, training or experience is required to conduct the procedure within this section.

10.6. Equipment

See Appendix 1 for example equipment checklist.

10.7. Procedure

10.7.1. Preparation for sampling

Prior to leaving for the field:

1. Ensure that all the field sampling equipment is in good order (**Figure 27**).
2. Ensure that the glass vacuum flask (**Figure 27**) is undamaged. Look for any signs of scoring, scratching and cracks and replace the flask if necessary.

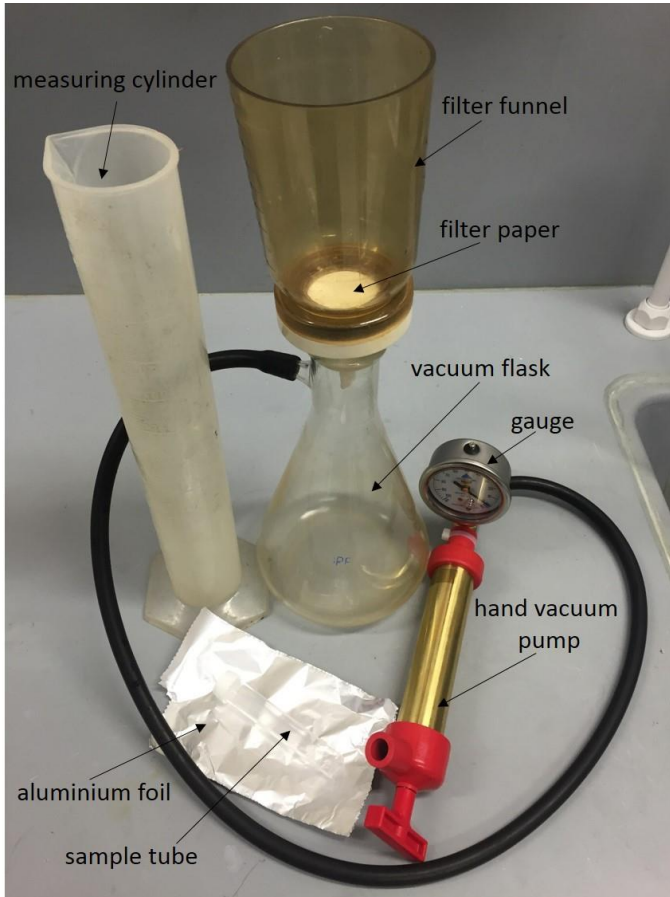


Figure 27: Vacuum filtration equipment

10.7.2. Field collection of water samples for chlorophyll a analysis

1. Set up the vacuum filtration equipment.
2. Secure the filter holder to the vacuum flask and confirm that the filter paper (**Figure 28**) is seated correctly.



Figure 28: Filter paper being placed into filter holder

3. Clear any floating matter from the water surface using the underside of the collection cylinder.
4. Invert the measuring cylinder and submerge into the water upside down to a depth of 0.2m so it fills with water.
5. Discard the excess water from the top of the cylinder to the uppermost 500mL or 100mL mark.
6. Pour a quantity of the collected water into the filter funnel, being careful not to spill any.
7. Pump the hand vacuum pump to start suction through the filter paper.
8. Pump until the vacuum gauge reads 40Kpa (1/2 atmospheric pressure) (or 5.8psi).
9. Maintain the pressure at 40Kpa throughout the filtering process to avoid possible rupture of cells and release of chlorophyll.
10. Continue to add known volumes of water to the filter for as long as the water flow through the filter is steady and the filter paper remains pale.
11. Refill the measuring cylinder when it is empty. Record the number of sample refills (cumulative volume).
12. Continue to add the water sample to the filter funnel.
13. Stop filtering if the vacuum flask is nearly overflowing. Remove the filter funnel from the flask, empty the flask, then reassemble the filter.
14. Stop filtering when the water flow through the filter has almost stopped and the filter paper has a noticeable colour (**Figure 29b**). Record the final volume filtered.
15. Remove the funnel from the filter holder.
16. Fold the filter paper in half over itself and remove it from the support screen.
17. Fold the filter paper again to reduce its size.
18. Place the folded filter paper into the correct sample tube and screw the lid on tightly.
19. Cover the sample tube completely with aluminium foil to exclude light.
20. Place sample tube into a labelled zip lock bag and place in cooler box. The bag should be labelled with the survey name and code, the survey date and the sampler's initials.
21. Ensure the sample name, site, date and time, sampler and GPS co-ordinates are recorded.
22. Record the total volume (in millilitres) of water that has been filtered through the filter paper. This information needs to be given to the analytical laboratory in order for them to calculate the concentration.

Note: If sample tubes are not available, filter paper may be double wrapped in aluminium foil and frozen.



(a)

(b)

Figure 29: (a) Algal bloom in the Caboolture River and (b) filter apparatus showing a well-covered filter paper for chlorophyll a analysis

10.7.3. Sample storage

After the survey, pack the zip lock bag containing all the chlorophyll samples (i.e. folded filter paper or tube, wrapped in aluminium foil) and put them in a freezer, ensuring samples are kept away from light. Recommended maximum holding time is four weeks.

When ready for analysis, remove the frozen chlorophyll samples from the freezer and transport them frozen to the analytical laboratory for analysis.

10.8. References and additional reading

Baird, RB, Eaton, AD, Rice, EW 2017, Standard Methods for the Examination of Water and Wastewater, 23rd edn, American Public Health Association, Washington DC, USA.

Appendix 1

Table 6: Equipment checklist

Equipment	✓
Note book or field sheet for recording data.	
500mL High Density Polyethylene (HDPE) plastic measuring cylinder	
100mL High Density Polyethylene (HDPE) plastic measuring cylinder	
1L ungraduated Buchner vacuum flask (side arm flask)	
Hand operated vacuum pump	
Vacuum gauge	
Silicon tubing to attach vacuum pump to flask	
Sample tubes (HDPE, 15mL volume, screw cap, graduated with 5 and 10mL levels), one tube per sample site	
0.4 micron glass fibre filter paper	
100mm x 100mm squares of aluminium foil	
One cooler box, two thirds filled with crushed ice to preserve samples	
Medium sized plastic zip lock bag to keep all the chlorophyll sampling tubes collected for each survey, marked with survey name, collection date and sampler's initials	
Test tube rack to hold chlorophyll sampling tubes ready for use	
Waterproof transport container for chlorophyll sampling equipment	
Powder-free disposable gloves	

Test kits for water sampling

Version: February 2018

11. Test kits for water sampling

11.1. Purpose and scope

This document provides background information on the use of test kits for water sampling.

11.2. Introduction

Test kits are used for rapid testing of water quality and can provide on-the-spot water quality information. However, test kits are often subject to a range of limitations and interferences and should be used with caution. Different brands and types of test kit can differ in a number of ways, such as in the level of accuracy, limit of detection, detection range and method used for the testing. The results produced by test kit-based analyses can also vary depending on whether they are being used to analyse freshwater, wastewater or saline waters. The most suitable test kit for each application should always be chosen and all instructions should be strictly followed.

Test kits should only be used in three situations:

- As a screening tool for the presence of an analyte of interest. The test kit must be capable of detecting the analyte at the environmentally significant concentration. If the analyte is detected, a water quality sample **must** be taken to determine a more reliable result through laboratory analysis.
- Where the holding time for an analyte is so short that it is not possible for the sample to be analysed by a laboratory (see example of chlorine below). In this case the test kit **must** be validated before use. It may be appropriate to undertake validation in conjunction with a National Association of Testing Authorities (NATA) accredited laboratory.
- As an emergency backup for instrument readings (for example, a damaged pH probe).

The performance of test kits must be verified to ensure they are fit for their intended purpose. Validation experiments covering, for example, the analytical range of the test kit should be undertaken before a particular test kit is used in the field. Validation should be undertaken in conjunction with a NATA accredited laboratory. Standards (if they are available) should be used to check results from a test kit each time it is used.

Key points regarding the use of test kits:

- Check the expiry date of the test kits and reagents and do not use them if it has passed.
- Store test kits correctly (for example, within the nominated temperature range, or out of direct sunlight, etc.).
- If possible, use the same test kit for the entire sampling program in order to reduce the risk of getting slightly different results from different test kits.
- If the test kit requires the same equipment to be used for each sample (for example, a tube to hold the water sample, or a dropper to transfer the sample), make sure it has been appropriately cleaned to minimise risk of cross-contamination.
- When using a test kit that relies on a colorimetric comparison to a reference chart, photograph the test solution against the colour chart (as in **Figure 30**).
- Water containing suspended particulate matter can potentially affect test kit results even at low concentrations, and so should be filtered appropriately before using the test kit.

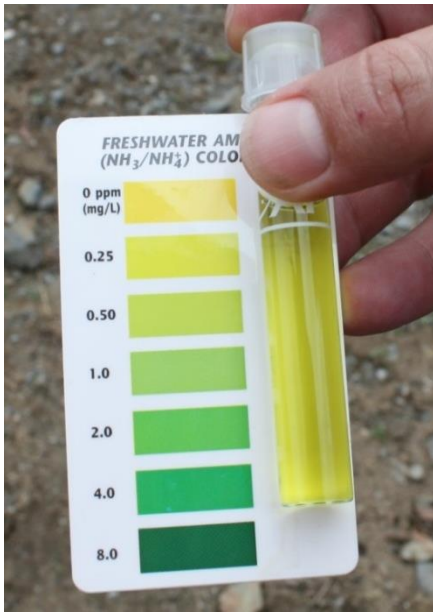


Figure 30: Freshwater ammonia test kit including tested water sample in vial and colour chart

11.3. Chlorine—an example of the suitable use of test kits

Chlorine is widely used for disinfection of public water supplies, swimming pools, and treated wastewaters. Some of the chlorine can be consumed in reactions with substances present in the water including ammonia, nitrite, and organic matter, producing chloramines and other disinfection by-products. Chlorination usually involves the addition of a measured dose of one or more of the following - chlorine gas (Cl_2), sodium hypochlorite (NaOCl), or hypochlorous acid (HOCl). The addition of chlorine gas alone results in a mix of all three in proportions dependent on factors such as pH and temperature.

The terms 'free chlorine' or 'free residual chlorine' are used to refer to the concentration of dissolved chlorine gas, hypochlorite ion, and hypochlorous acid left after the initially added chlorine has reacted with water constituents. The term 'combined chlorine' refers to chloramines produced by the reaction of chlorine with (most commonly) ammonia, and the terms 'total chlorine' or 'total residual chlorine' refers to the sum of 'free chlorine' and 'combined chlorine'. Because the levels of 'free chlorine' relative to 'combined chlorine' can change over a very short period of time, it is usual to measure chlorine using a test kit or probe attached to a water quality meter. Test kits commonly used for chlorine testing are based on a colorimetric system, involving the addition of a chemical DPD (N,N-diethyl-p-phenylenediamine), typically supplied in the form of tablets or powder in a sealed container, to be applied to a prescribed volume of sample water. The intensity of pink colour produced by the reaction of the DPD with the chlorine present in the water is then measured.

The methods used to measure the colour intensity vary between test kit types. Simple kits involve comparison by eye of the colour intensity with a calibrated chart or filter (similar to the ammonia chart illustrated in **Figure 30**). More sophisticated (and accurate) methods measure colour intensity digitally, using a handheld spectrophotometer. The results from DPD-based test kits may be adversely influenced by colours and interferences from chemicals present in the waters being tested.

Background information on the collection and preservation of sediment

Version: February 2018

12. Background information on the collection and preservation of sediment

12.1. Purpose and scope

This document describes important considerations prior to undertaking a sediment sampling plan. The associated document Collection and preservation of sediment outlines steps to be undertaken when collecting sediment.

Objectives **not** covered in this manual include:

- Where sediments are to be sampled as part of an evaluation of the suitability of dredge sediments for ocean or land disposal. Information of sediment sampling requirements for dredging activities can be obtained from the National Assessment Guidelines for Dredging (Commonwealth of Australia 2009). Information on appropriate sampling equipment to be used to collect samples for this purpose can be obtained from the U.S. EPA (2001) Methods for Collection, Storage and Manipulation of Sediments for Chemical and Toxicological Analyses: Technical Manual.
- Where sediments are to be used as part of a sediment toxicity assessment. Methods relating to the collection of sediments for this purpose are presented elsewhere in this manual (see Direct toxicity assessments).

12.2. Associated documents

Physical and chemical assessment:

- *Collection and preservation of sediment*
- *Manual collection of surface water samples (including field filtration)*

Sampling design and preparation:

- *Background information on water quality measurements using in situ water quality instruments*
- *Water quality sampling using in situ water quality instruments*

Biological assessment: Direct toxicity assessments

12.3. Introduction

The analysis of inorganic and organic contaminants in sediment allows for the assessment of the risk to aquatic or benthic ecosystems or other environmental values. Sediment samples are typically more heterogeneous (poorly mixed) than water and wastewater samples, and as such, care must be taken when designing a sediment sampling plan to ensure the sample is representative of the environment being assessed.

12.4. Considerations for sediment sampling

In order to collect a representative sample, there are a number of considerations that need to be made. The exact sampling design will depend upon many factors, such as the purpose of the sampling, the type of aquatic environment and sediment composition to be sampled, site access and availability of equipment. More detailed information on sediment sampling and considerations are available in Simpson and Batley (2016).

Although the collection site may be predetermined (e.g. to correspond to water sample collection, or ease of access), sediments should be collected from locations where finer sediments tend to be deposited. This is because fine sediments offer a greater surface area for adsorption of contaminants, and also because fine sediments generally contain higher proportions of organic matter that can readily bind metallic and organic compounds. Sediments with a higher proportion of finer grains can typically be found where water current velocities are low (e.g. large and deep water-holes, inside bends in rivers).

Sediment is typically heterogeneous both horizontally and vertically. Therefore, care must be taken to ensure replication and/or composite sampling is sufficient to obtain a representative sample from the potentially impacted area of sediment being assessed. For example, a composite sample can be made up with multiple samples collected one to ten metres apart and combined by thorough mixing. Alternatively, where disturbance of sediments prior to testing needs to be minimised (e.g. for analysis of volatile substance or where reduction/oxidation (REDOX)

conditions need to be maintained), multiple samples may be collected for testing. Ideally, the number of replicate samples required, the depth at which they are taken, and the distance between sampling sites should be defined prior to collection.

12.4.1. Holding times and preservation

Sediment samples must be delivered to the laboratory to allow for analyses or tests to be conducted within the prescribed holding and testing times. Holding times will vary depending on the analyses to be performed and are dictated by the compound-specific analysis requirements. Sediment toxicity tests should be commenced as soon as practical after sample collection and two weeks is recommended as a maximum holding time. A summary of the preservation and holding times for various sediment analyses is presented in Table 1; however, it is best practice to seek advice on preservation and holding times from the laboratory that will be conducting the analysis.

Table 1: General guide to preservation and holding times for sediment analyses

Analysis	Preservation	Holding Time
Pore-waters	For metals, consult testing laboratory for specific instructions. For dissolved metals, filter (0.45 µm) and acidify immediately after pore-water extraction. If oxidation state of metal is likely to affect solubility, extraction and preservation should be performed in an inert atmosphere (e.g. under nitrogen). For organic contaminants: refrigerate at <6°C. For ammonia or sulfide, see testing laboratory for instructions.	Analyse samples that may contain volatile analytes (ammonia, sulfide and organics) as soon as possible after collection. Analyse preserved metal samples within 14 days.
Whole sediments for total particulate metals (with exception of mercury)	None	Six months.
Whole sediments for mercury	Refrigerate at <6°C and store in the dark.	28 days.
Whole sediments for bioavailable metals or for metal speciation.	Refrigerate at <6°C, unless metals associated with acid volatile sulfide (AVS) are the contaminants of interest, in which case freezing is appropriate.	Analyse as soon as practical, but generally within 7 days.
Whole sediments for extractable organics. Note: Total Organic Carbon should always be measured when sampling for organics.	Refrigerate at <6°C and store in the dark.	Analyse within seven days for volatile compounds or those prone to microbial degradation. Analyse within eight weeks for stable organics such as organochlorine pesticides, dioxins/furans, polychlorinated biphenyls (PCB), polycyclic aromatic hydrocarbons (PAH).
Whole sediments for acid volatile sulfide (AVS), ammonia or other volatile inorganic compounds	Frozen	28 days
Where sediments are to be used for toxicity assessments, holding times and sample storage conditions should meet the requirements described for the specific compounds present (with appropriate refrigeration; <6°C), but generally no longer than two weeks.		

12.5. References and additional reading

AS/NZS 5667.12:1999, Water quality—Sampling—Guidance on the Sampling of Bottom Sediments.

Commonwealth of Australia 2009, National Assessment Guidelines for Dredging, Commonwealth of Australia, Canberra, 81pp.

Simpson, SL, and Batley, GE 2016, Sediment quality assessment: A Practical Handbook, 2nd edn, CSIRO Publishing, Canberra, 360pp.

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Collection and preservation of sediment

Version: February 2018

13. Collection and preservation of sediment

13.1. Purpose and scope

This document describes methods for the collection and preservation of sediments for the purpose of undertaking analysis of inorganic and organic contaminants. This will assist in an assessment of the risk to aquatic or benthic ecosystems or other environmental values. This document must be read in conjunction with the associated document Background information on the collection and preservation of sediment, which provides important considerations that must be made prior to determining and conducting a sediment sampling plan.

13.2. Associated documents

Physical and chemical assessment:

- *Background information on the collection and preservation of sediment*
- *Water quality sampling using in situ water quality instruments*
- *Sampling design and preparation*
- *Permits and approvals*
- *Operating a basic handheld GPS unit for an investigation of compliance inspection*
- *Record keeping including taking field photographs and videos*

Biological assessment: Direct toxicity assessments

13.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

13.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals for more information on requirements.

13.5. Skills, training and experience

Skills, training or experience required to conduct the procedure within this section include experience in using the equipment described in the method.

13.6. Equipment

See Appendix 1 for example equipment checklist.

13.7. Procedure

13.7.1. Preparation for sampling

1. Determine the sampling procedure prior to sampling, giving consideration to:
 - 1.1. The exact location for sample collection, ensuring it is representative of the site.
 - 1.2. Whether a composite sample, multiple replicate samples or a single discrete sample is appropriate to obtain a representative sample of the site.
 - 1.3. The number of replicate samples (if required), the depth at which they are taken, and the distance between replicate sampling sites prior to collection.
 - 1.4. If collecting a composite sample, it can be made up with multiple samples collected one to ten metres apart and combined by thorough mixing. However, where disturbance of sediments prior to testing needs to be minimised (i.e. no mixing is possible), multiple replicate samples should be taken.
2. Prepare any sampling equipment that may be required. Stainless-steel equipment is generally appropriate for sampling sediments; however, if ultra-trace metal concentrations are to be sampled it would be appropriate to use plastic (HDPE or PTFE) sampling equipment.
 - 2.1. Clean all sampling equipment to be used with a laboratory grade detergent and rinse three times with laboratory grade distilled water. Wrap in aluminium foil or clean heavy duty plastic to keep dust free.
 - 2.2. If sampling for organics, wipe equipment down with an organic solvent (such as laboratory grade hexane) and wrap equipment in aluminium foil. Hexane is preferred for analysis of petroleum hydrocarbons.
 - 2.3. If sampling for ultra-trace metals, rinse equipment in 10% nitric acid.
3. Notify the analysis laboratory prior sampling as to whether sieving is necessary and if analysis of sub-fractions is required. Laboratory analysis may require sieving to obtain sub-fractions, with <math><2000\mu\text{m}</math> or <math><63\mu\text{m}</math> being most common. You will need to inform the laboratory if you require analysis of sub-fractions. Discuss storage requirements and holding times if unsure.

Note: It is essential that all sources of contamination are excluded from the sampling process, including residues from tobacco products, food and beverage, sunscreen lotions, cosmetics and cleansers, water and sediments carried on clothing and equipment.

13.7.2. Sampling

1. Label sampling containers (e.g. jars, bags) to be used.
2. Clean all sampling equipment prior to sampling as in Step 2.1 above. Use of field water for cleaning equipment may also be appropriate for relatively clean environments.
3. Put on clean gloves, ensuring the type corresponds to the chemical risk.
4. Either wade or navigate the boat (depending on type of sampling) to the sampling location to collect sediments, taking care not to disturb sampling area.
5. Find a suitable sampling location, aiming to collect sediments from locations where finer sediments tend to be deposited (i.e. at sections with low water velocities such as large and deep water-holes or inside bends in rivers).
6. Collect sediments using a suitable sampling device (such as a clean trowel, Van Veen Grab Sampler or corer).
7. Siphon off any overlying water with care to minimise loss of fine surface sediments that may be resuspended.
8. Take a photo of the sample.
9. If sub-sampling from a core sample, specific depth horizons can be selected by extruding the core and cutting the exposed sediment with a stainless steel or plastic (HDPE or PTFE) cutter.
10. If collecting a composite sample, place sediment from different individual samples into a stainless steel or plastic (HDPE or PTFE) bowl. If possible, avoid using sediment that has been in direct contact with the metal sampling device. Thoroughly mix the sediment in the bowl using a stainless steel or plastic (HDPE or PTFE) spoon or other tool before sub-sampling.

Note: It is necessary to consider whether mixing will influence the results for the contaminants being analysed. See Background information on the collection and preservation of sediment document for more information.

11. Place the sediment into the individually labelled sample container/s.
12. If the sample IS NOT to be frozen:
 - 12.1. Fill the container almost to the brim and cover with water from the collection site leaving no airspace.
 - 12.2. Seal the container with an appropriate lid that will prevent leakage or minimise ingress of air.
 - 12.3. If sediment oxidation is a concern, the headspace or cover-water can be purged with an inert gas (e.g. nitrogen or argon). Bubble gas (via a narrow tube connected to the cylinder) through the

overlying water to drive off oxygen and then replace the lid tightly.

12.4. Store the samples vertically to avoid mixing.

12.5. Keep cool or refrigerate promptly.

13. If the sample is to be frozen e.g. for measurement of acid volatile sulfide (AVS):

13.1. Fill the container to only two-thirds of capacity, including any cover water taken from the same site.

13.2. Store the samples vertically.

13.3. Place samples promptly in dry ice or portable freezer.

14. If possible, measure in situ water quality data 5 to 20cm above the sampling site (i.e. pH, electrical conductivity, temperature, redox potential, turbidity and dissolved oxygen).

15. Collect sediment quality characteristics if required (e.g. pH, redox potential).

16. Record all relevant information relating to the sample collected in the field notebook/field data sheets, including:

- date, time, site name, GPS coordinates
- sediment collection method (i.e. use of sampling device, composite sampling etc.)
- water column depth above sediment collection site
- sampling conditions (water depth, tides, waves, water clarity).

13.7.3. Post sampling

Sediment samples must be delivered to the laboratory to allow for analyses or tests to be conducted within the prescribed holding and testing times. Holding times will vary depending on the analyses to be performed.

Sediment toxicity tests should be commenced as soon as practical after sample collection and two weeks is recommended as a maximum holding time. See Direct toxicity assessments for further information.

A summary of the holding times for various sediment analyses is presented in Table 1 of the Background information on the collection and preservation of sediment document.

13.8. References and additional reading

AS/NZS 5667.12:1999, Water quality—Sampling—Guidance on the Sampling of Bottom Sediments.

Commonwealth of Australia 2009, National Assessment Guidelines for Dredging, Commonwealth of Australia, Canberra, 81 pp.

Simpson, SL, and Batley, GE 2016, Sediment quality assessment: A Practical Handbook, CSIRO Publishing, Canberra, 360 pp.

U.S. EPA 2001, Methods for Collection, Storage and Manipulation of Sediments for Chemical and Toxicological Analyses: Technical Manual, Office of Water, United States Environmental Protection Agency, Washington, DC, 208 pp.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Single or multi-parameter water quality instrument	
Field data sheets, notebook or field computer	
Labelling equipment (e.g. stickers and/or permanent markers)	
Personal Protective Equipment (PPE): <ul style="list-style-type: none"> • Safety boots, high visibility vest, long pants, long-sleeved shirt, hardhat (industrial sites); • Waders, gumboots, broad-brimmed hat or similar PPE (shore-based sampling). 	
Gloves - non-powdered latex free (e.g. nitrile) corresponding to the chemical risk	
Clean collection containers as provided by the analytical laboratory	
Clean sediment sampling equipment* for collection of surficial layers (<10cm depth): <ul style="list-style-type: none"> • stainless steel (organics or metals) or plastic (HDPE or PTFE for ultra-trace metals) trowel for dry sediments or for exposed littoral sediments (tidal flats); • stainless steel benthic grab sampling equipment (e.g. Ponar or Van Veen) grab sampler for deployment from a boat or jetty. 	
Clean hand corer for sediment profiling	
Clean stainless steel sieve (10mm) for removal of stones, plant material or fauna	
Cool-box with ice bricks or portable refrigerator	

* For further information of appropriate sampling devices see U.S. EPA (2001).

Guidance on the sampling of groundwaters

Version: May 2018

14. Guidance on the sampling of groundwaters

14.1. Purpose and scope

This document provides some general information on groundwater and its importance for assessment of impacts. Detailed instructions on how to sample groundwaters are provided in Geosciences Australia Groundwater sampling and analysis – a field guide (Sundaram et al. 2009), and AS/NZS 5667.11:1998 (R2016), Water Quality – Sampling – Guidance on sampling of groundwaters. Both of these documents should be referred to when undertaking groundwater sampling, and the Geosciences Australia field guide (Sundaram et al. 2009) can be used as a default standard operating procedure (SOP) for groundwater sampling.

14.2. Introduction

Groundwater is a valuable natural resource that has a range of environmental values including the provision of drinking water for humans and livestock, cultural and spiritual values, ecosystem values and provision of water flows to groundwater dependent ecosystems. Groundwater is also used for agriculture and industrial applications. In dryland areas, groundwater can be the only reliable source of water and can sustain water levels in river and wetland ecosystems during extended dry periods. In Queensland, the Environmental Protection Act 1994 and the Environmental Protection (Water) Policy 2009 provide a framework for protecting groundwater quality.

Groundwater quality can be highly variable, both spatially and temporally (Australian Government 2013), more so than surface water quality. Groundwater quality can be influenced by local geology, residence time in the aquifer, groundwater chemistry and groundwater-rock interactions. Groundwater can have naturally elevated salinity concentrations, dissolved nutrients and metals.

14.3. Understanding the system

The assessment of impacts to groundwater requires an understanding of the aquifer system that hosts the groundwater, including its depth, physical and chemical properties, and hydrologic interaction with surface water systems. The term 'groundwater' refers to water that seeps into the ground and accumulates in the pores and cracks of the saturated zone of the earth's crust. The term "aquifer" refers to the body of rock or sediment that can store and transmit groundwater. Groundwater can occur in the saturated zone where all available spaces are filled with water, and in the unsaturated zone, the space between the land surface and the water table (upper surface of the zone of saturation of an unconfined aquifer), where there are pockets of air that contain some water (Centre for Groundwater Studies 2001).

If an aquifer becomes polluted, the polluted water can be transported to the surface at one or more discharge sites. This can occur naturally via normal discharge (e.g. to springs, creeks, wetlands) or artificially via extraction/abstraction from a bore or pumping well. This is important when the discharge site is a surface water body that is used for drinking water, agriculture (i.e. livestock watering), industrial applications, or has aquatic ecosystem values.

Poor groundwater quality has the potential to impact extractive users of the groundwater, surface water quality and groundwater dependent ecosystems (GDE). A GDE is an ecosystem that requires access to groundwater to maintain communities of plants and animals, ecological processes and ecosystem services. GDEs can depend on sub-surface and surface expression of groundwater and can be categorised into three broad types (Australian Government 2013):

- Surface ecosystems that rely on groundwater discharge to rivers, wetlands and springs
- Surface ecosystems that access groundwater from below the water table, e.g. terrestrial vegetation
- Subterranean aquatic ecosystems, which include stygofauna, in aquifers and caves.

14.4. Groundwater sampling

Groundwater sampling requires special equipment for sampling from a bore or, with a procedure to ensure sampling of fresh and undisturbed groundwater or water representative of the aquifer, as opposed to potentially stagnant water held in the bore column. Sometimes special precautions are needed to prevent changes in quality of the groundwater due to effects such as:

- reduced pressure when brought to the surface. This can cause gases in solution at the higher pressures found

underground to move into a gas phase at the surface. In some cases, these can be toxic gases, such as hydrogen cyanide if the groundwater has been contaminated by cyanide solution.

- exposure to components of the atmosphere such as oxygen. This can oxidise compounds naturally present in the reduced form (for example, ferrous ions).
- temperature changes can influence the kinetic energy (chemical) of the system. This can influence the redox reaction regime and the rate of bio-degradation. Temperature changes could also increase the volatilisation of dissolved constituents.

Detailed information on collecting groundwater samples is provided in Geosciences Australia Groundwater sampling and analysis – a field guide (Sundaram et al. 2009) and AS/NZS 5667.11:1998 (R2016) Water Quality – Sampling – Guidance on sampling of groundwaters. More detailed information on low flow sampling can be found in USEPA (1996). Sampling should be undertaken in line with these documents.

Purging or low flow sampling methods are preferred for accurate groundwater sampling. Low flow methods minimise the impact of the sampling method on the aquifer and are more likely to obtain a representative sample, while some high flow pumps can sometimes induce water chemistry changes. Bailers: should only be used where no other pump can be used; b) should only be used for shallow wells where a small volume of water needs to be purged prior to sampling; and c) water samples taken using a bailer may have lower accuracy for some analytes and therefore it is important to record that a bailer was used to collect water samples.

14.5. Groundwater quality assessment

Groundwater quality is generally assessed based on a comparison of measured groundwater quality indicators against guideline values. Groundwater investigation levels (GILs) are provided in Schedule B6 of the National Environment Protection (Assessment of Site Contamination) Measure 1999 (April 2013). A framework for risk-based assessment of groundwater that has been affected, or may have been affected by site contamination is also provided in this document. Basin specific water quality objectives (WQO) may be prescribed under the Environmental Protection (Water) Policy 2009 (see <https://www.ehp.qld.gov.au/water/policy/>).

It is essential that the groundwater system and groundwater quality characteristics are adequately described and understood in order to assess current groundwater quality and the potential future risks to the groundwater. Shallow groundwater quality in particular can be variable temporarily and spatially due to a range of factors including changes in the lithology of the aquifer, seasonal conditions, recharge events, and pumping regimes.

The information required would typically include:

- descriptions of groundwater hydrogeology and hydraulics within the potentially impacted aquifer/s
- water quality characteristics, of the groundwater within the potentially impacted aquifer/s (including the major cation and anion composition).

14.6. References and additional reading

AS/NZS 5667.11:1998 (R2016), Water Quality – Sampling – Guidance on sampling of groundwaters, Standards Australia.

ANZECC & ARMCANZ 1998, National Water Quality Management Strategy. Implementation Guidelines. Australian and New Zealand Conservation Council, Agriculture Resource Management Council of Australia and New Zealand, Commonwealth of Australia, Canberra.

Australian Government 2013, Guidelines for Groundwater Quality Protection in Australia - 2013. National Water Quality Management Strategy, Australian Government, Canberra.

Centre for Groundwater Studies 2001, Australian Groundwater School Fundamentals of Groundwater Science, Technology and Management, 3rd ed.

Department of Environment and Heritage Protection (DEHP) 2010, Queensland Water Quality Guidelines, Version 3, ISBN 978-0-9806986-0-2, Department of Environment and Heritage Protection (DEHP), Queensland Government, Brisbane.

Department of Science Information Technology and Innovation (DSITI) 2017, Using monitoring data to assess groundwater quality and potential environmental impacts, Version 1, Department of Science, Information Technology and Innovation (DSITI), Queensland Government, Brisbane.

National Environment Protection (Assessment of Site Contamination) Measure 1999 (April 2013), NEPC 2013, Canberra.

Sundaram, B, Feitz, A, Caritat, P de, Plazinska, A, Brodie, R, Coram, J and Ransley, T 2009, Groundwater

Sampling and Analysis – A Field Guide, Geosciences Australia, Record 2009/27. Available from:
<http://www.cffet.net/env/uploads/gsa/BOOK-Groundwater-sampling-%26-analysis-A-field-guide.pdf>

United States Environmental Protection Agency (USEPA) 1996, Low stress (low flow) purging and sampling procedure for the collection of ground water samples from monitoring wells. Available from:
https://www.orau.org/ptp/PTP%20Library/library/Subject/Environmental/r1_lowflow.pdf

15. Swab sampling

15.1. Purpose and scope

This document provides a method for swab sampling.

Swab (or wipe) sampling can be used to detect organic and inorganic contaminants (dusts, pesticides, metals, spray drift, contaminant residues, etc.) on different surfaces. The technique is most effective on smooth surfaces such as glass, metal (including pipes), painted surfaces and smooth vegetation surfaces such as leaves. Swab sampling is less effective on surfaces that are rough and/or porous (e.g. timber and concrete).

Although there are regulatory measures of surface contamination for some contaminants (e.g. for 'PCB-free' materials), the use of wipe samples for a purpose other than detecting contaminant presence is not recommended.

15.2. Associated documents

Sampling design and preparation: Record keeping, including taking field photographs and videos

15.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

15.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

15.5. Skills, training and experience

No skills, training or experience is required to conduct the procedure within this section.

15.6. Equipment

See Appendix 1 for example equipment checklist.

15.7. Procedure

15.7.1. Choosing the material for swab sampling

There are a variety of commercially available swab materials. Contact your laboratory supplier for recommendations on a suitable choice of swab material. When there is no time to obtain laboratory grade swabs before sampling, there are readily available materials that are suitable for use as swabs such as filter papers, small gauze pads and cotton tips. Portions of these material chosen should be uniform in all respects and free of measurable contamination by the analytes of interest (this must be checked by analysing blank swabs). There is a higher possibility of contamination and matrix interferences using readily available materials, and therefore their use is only recommended for urgent sampling when only the presence or absence of a contaminant is analysed for.

Analytical grade solvent should be used to wet the swab or wipe. Organic solvents, such as isopropyl alcohol or hexane, are generally used. Ultrapure water is only an appropriate wetting solvent for inorganic dusts. Contact your laboratory for advice on suitable solvents for the analyte of interest.

15.7.2. Taking the swab sample

1. Choose a standard area to swab (minimum of 10cm²). Record the standard area.
2. Clean, powder-free gloves must be worn.
3. Pre-mark the standard area on the surface to be wiped. Caution should be used when pre-marking a surface. For example, pre-marking a surface with permanent marker and then using an organic solvent may lead to the

permanent marker ink dissolving into the solvent and causing contamination. Alternatively, a pre-cut template held against the surface can be used.

4. Wet each swab with appropriate solvent. Record the solvent used.
5. Wipe the swab across the pre-marked surface from left to right using an even pressure and holding the swab flat against the surface.
6. Continue until the whole surface has been wiped (**Figure 31a**).
7. Re-wipe again top to bottom (**Figure 31b**).
8. Re-wipe again bottom left to top right (**Figure 31c**).
9. Re-wipe again top left to bottom right (**Figure 31d**).
10. Used swabs should be sealed in labelled sampling containers appropriate for the storage of the analytes of interest (e.g. a solvent-washed glass jar if pesticides are the contaminant of interest). Record the sample name, site, date and time and sampler in a notebook or equivalent.
11. If a template has been used, it must be cleaned appropriately before using it at another site.
12. A field blank should be taken by wetting the swab/s with the solvent and placing the swab in the jar.

When only trace levels of contamination are anticipated, a larger surface area should be swabbed (up to 1m²). More than one wipe may be used, and wipes can be pooled for analysis.

If plant leaves are to be swab sampled, a uniform area could be approximated by swabbing the surfaces of a fixed number of leaves of similar size at each site.

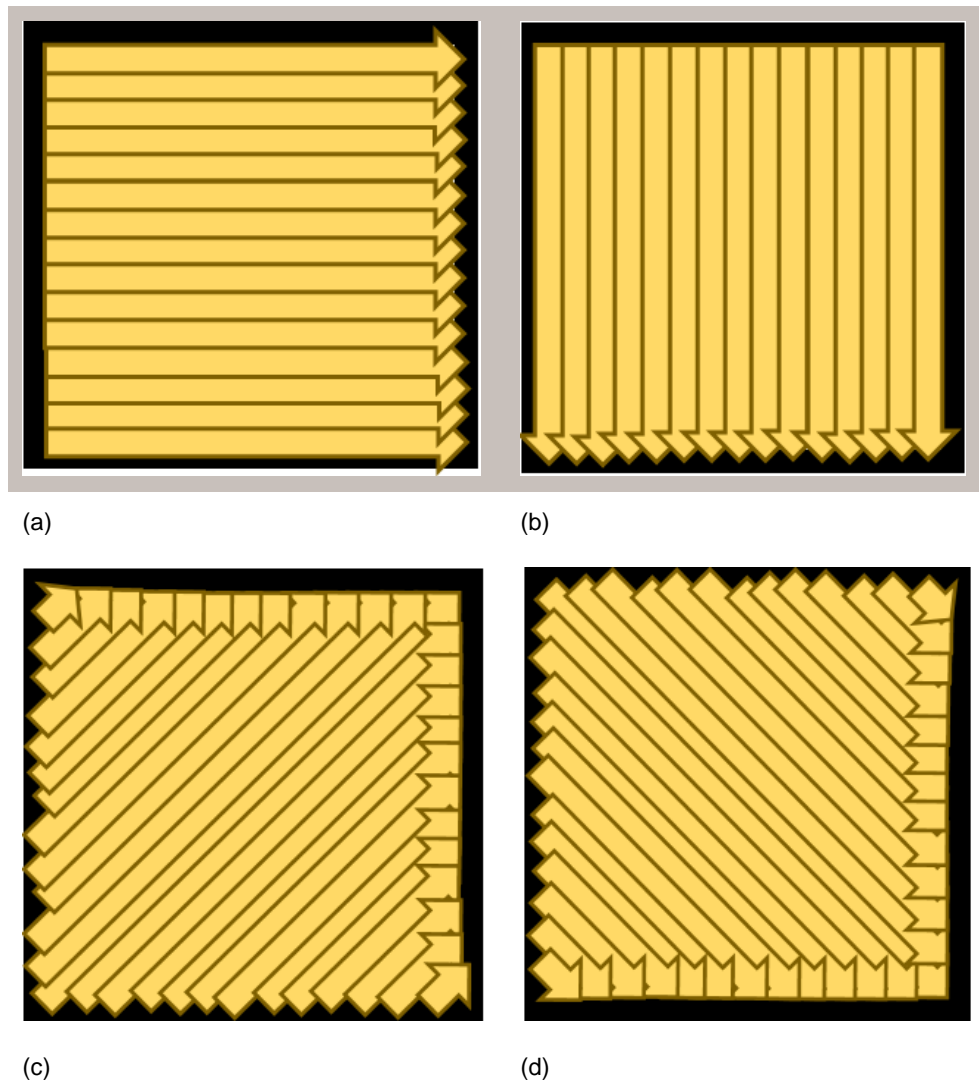


Figure 31: Example of procedure for swab sampling, with black outline indicating the pre-marked area to be swabbed, yellow arrows indicating the direction for swiping. Swabbing starts at a) swab the area from left to right, b) swab the area from top to bottom c) swab the area from bottom left to top right, and d) swab the area from top left to bottom right.

Appendix 1**Table 1: Equipment checklist**

Equipment	✓
Field sheet or note book for recording data.	
Clean, powder-free gloves	
AR grade solvent of choice	
Swabbing material of choice	
Labelled sample containers made from a material appropriate to the contaminant of interest	
Commercial test kits including pre-cut templates if required	

Background information on stable isotope analyses

Version: February 2018

16. Background information on stable isotope analyses

16.1. Purpose and scope

This document provides background information on stable isotope analyses.

16.2. Associated documents

Physical and chemical assessment: Sampling and preparation for stable isotope analyses of biota and sediment

Refer also to other relevant documents within this manual, which outline methods for:

- sediment sampling
- fish sampling
- macroinvertebrate sampling but **DO NOT preserve the samples with ethanol**
- chlorophyll a sampling.

16.3. Introduction

Isotopes are atoms of the same element, each having a different number of neutrons. Carbon, hydrogen, oxygen, nitrogen and sulphur comprise the bulk of living bodies (along with phosphorus) and all have more than one stable (i.e. non-radioactive) isotope. Stable isotopes occur naturally in the environment and can (amongst other things) be used by aquatic ecologists to track elements such as carbon and nitrogen through a food chain.

Analysis of stable isotopes ratios provide two types of information:

- **fractionation information** – where stable isotopes are fractionated (the ratio of isotopes are changed) through chemical reaction, diffusion or evaporation, and the ratio reflects the reaction, diffusion or evaporation conditions
- **source information** – where stable isotope ratios are used to provide information about origin of a sample (Peterson and Fry, 1987). The source sets an isotopic baseline that can subsequently be shifted by isotopic fractionation.

Stable isotopes can therefore be used in studies of aquatic ecosystems and when investigation surface and groundwater systems to answer questions related to:

- plant and animal ecophysiology
- trophic structure
- energy pathways within ecosystems and at their interfaces (e.g. with terrestrial environments and between freshwater and marine environments)
- hydrology and hydrogeology.

16.4. Food chains

The use of isotopes to trace nutrient and energy sources and understand trophic interactions depends on variations in isotope ratios in organisms and in their environment. The variation in isotopic ratios of a food source and the consumer of that food source will determine if, how, and when stable isotope techniques may be applied (Michener and Lajtha, 2007), for example:

- The heavier carbon isotope ^{13}C is one of the most effective tracers of organic carbon sources and energy flow in aquatic ecosystems because it does not change much as it is transferred up the food chain, following fixation by plants and undergoes minimal changes during decomposition (Michener and Lajtha, 2007; Tiunov, 2007).
- Sulphur isotopes can also be used for determining food sources, especially in coastal environments or ecosystems with strong gradients in redox conditions (Michener and Lajtha, 2007).
- Nitrogen stable isotopes are a powerful tracer of the nitrogen cycle. The ratio of heavy (^{15}N) to light (^{14}N) nitrogen isotopes changes markedly from food source to consumer and can be used to understand food web interactions in aquatic ecosystems.

16.5. Hydrology and hydrogeology

Stable isotopes can also be used in hydrology and hydrogeology studies to understand the surface-subsurface

water interactions including:

- dominant runoff producing processes
- geographic source of water
- recharge sources or pathways
- the origins of water and solutes and weathering pathways
- flow within and between aquifers and surface waters
- residence time of water in the subsurface (Michener and Lajtha, 2007).

There are many stable isotopes of different elements that may be used in hydrology and hydrogeology studies. Their value to an investigation depends on the purpose of the investigation and the questions it aims to address. When using stable isotopes in hydrology and hydrogeological studies, care should be taken to consider the application of different stable isotopes to the investigation and the ability for particular stable isotopes to address particular issues relevant to the investigation.

16.6. Sediment source finger-printing

Stable isotopes can also be used in aquatic ecosystem sediment source fingerprinting (Fox 2009; Fox et al. 2010; Laceby et al. 2015; Walling 2013). This involves collecting a sample of the sediment transported or deposited in an aquatic ecosystem and comparing its stable isotope composition with that of potential sediment sources within the catchment area. In this way it is possible to gain information on the relative importance of different potential sediment sources. For example:

- Stable isotopes of carbon and nitrogen, which are linked specifically to the organic fraction of sediment, can discriminate soils from:
 - areas under different vegetation cover (C₃ versus C₄ vegetation)¹¹
 - surface soils or from subsurface soils
 - different nutritional contributions of aquatic organisms to the organic components of the soil (Marwick et al. 2014; Nadelhoffer and Fry 1988; Tiunov 2007).
- Stable isotopes can be used to trace the source of the organic component of suspended sediment and help to identify the source of the sediment (Bellanger et al. 2004; Garzon-Garcia 2014; Gomez et al. 2010; Kao and Liu 2000).

The use of compound-specific isotope analysis (CSIA) of carbon and hydrogen associated with plant fatty acids and long-chain n-alkanes has more recently proven to be useful in discriminating soils from areas under different vegetation (Cooper et al., 2015; Hancock and Revill, 2013).

16.7. References and additional reading

Bellanger, B, Huon, S, Velasquez, F, Valles, V, Girardin, C and Mariotti, A 2004, 'Monitoring soil organic carbon erosion with d¹³C and d¹⁵N on experimental field plots in the Venezuelan Andes', *Catena*, 58, 125-150.

Cooper, RJ, Pedentchouk, N, Hiscock, KM, Disdle, P, Krueger, T and Rawlins, BG 2015, 'Apportioning sources of organic matter in streambed sediments: An integrated molecular and compound-specific stable isotope approach', *Science of the Total Environment*, 520, 187-197.

Fox, JF 2009, 'Identification of sediment sources in forested watersheds with surface coal mining disturbance using carbon and nitrogen isotopes', *American Water Resources Association*, 45 (5), 1273-1289.

Fox, JF, Davis, CM and Martin, DK 2010, 'Sediment source assessment in a lowland watershed using nitrogen stable isotopes', *Journal of the American Water Resources Association*, 46 (6), 1192-1204.

Fry, B 2006, *Stable Isotope Ecology*. Springer, New York.

Garzon-Garcia A, Laceby JP, Olley JM and Bunn SE 2017, 'Differentiating the sources of fine sediment, organic matter and nitrogen in a subtropical Australian catchment', *Science of the Total Environment*, 575, 1384-1394.

Gomez B, Baisden WT and Rogers KM 2010, 'Variable composition of particle-bound organic carbon in steepland river systems', *Journal of Geophysical Research-Earth Surface*, 115.

¹¹ C₃ and C₄ refers to a certain photosynthetic pathway. Most plants are C₃ plants. C₄ plants include some grasses, sugar cane, maize and sorghum.

- Hancock GJ, Revill AT 2013, 'Erosion source discrimination in a rural Australian catchment using compound-specific isotope analysis (CSIA)', *Hydrological Processes*, 27, 923-932.
- Kao SJ, Liu KK 2000, 'Stable carbon and nitrogen isotope systematics in a human disturbed watershed (Lanyang-Hsi) in Taiwan and the estimation of biogenic particulate organic carbon and nitrogen fluxes', *Global Biogeochemical Cycles*, 14, 189-198.
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- Michener, R and Lajtha, K 2007, *Stable Isotopes in Ecology and Environmental Science*, 2nd edn, Blackwell Publishing, Singapore.
- Nadelhoffer KJ and Fry B 1988, 'Controls on natural nitrogen-15 and carbon-13 abundances in forest soil organic matter', *Soil Science Society of America Journal*, 52, 1633-1640.
- Peterson BJ and Fry B 1987, 'Stable isotopes in ecosystem studies', *Annual Review of Ecology and Systematics* 18, 293-320.
- Tiunov, AV 2007, 'Stable isotopes of carbon and nitrogen in soil ecological studies', *Biology Bulletin*, 34, 395-407.
- Walling, DE 2013, 'The evolution of sediment source fingerprinting investigations in fluvial systems', *Journal of Soils and Sediments*, 13, 1658-1675.

Sampling and preparation for stable isotope analyses of biota and sediment

Version: February 2018

17. Sampling and preparation for stable isotope analyses of biota and sediment

17.1. Purpose and scope

This document outlines the methods for sample collection, preservation and storage of samples prior to transport and delivery to a laboratory for carbon or nitrogen Stable Isotope Analysis (SIA). It does not cover collection of water samples.

Methods described in this document should be discussed with the analytical laboratory staff prior to sample preparation in order to determine specific requirements for that laboratory. The methods have been adapted from the Stable Isotope Analysis Protocol from the Australian Rivers Institute, Griffith University.

Further information on methods and issues associated with SIA are provided in Section 17.8. These documents should be consulted prior to developing a SIA monitoring program.

17.2. Associated documents

Physical and chemical assessment: Background information on stable isotope analyses

Sampling design and preparation – Record keeping including taking field photographs and videos

Refer also to other relevant documents within this manual, which outline methods for sediment sampling, fish sampling, macroinvertebrate sampling (but **DO NOT preserve the samples with ethanol**) and chlorophyll a sampling.

17.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

17.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Note: Some crustaceans (e.g. crabs, prawns and lobsters) are considered fish under the Fisheries Act 1994 and therefore relevant permits and approvals need to be sought.

If a boat is to be used for research and/or commercial purposes, consideration must be given as to whether a Certificate of Survey or Marine Safety (scientific research and educational activities) Exemption is required.

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals for more information on requirements.

17.5. Skills, training and experience

Staff using this method should have previous training and experience in sampling protocols specific to sediments, plants, algae or fish depending on which sample type is to be used.

Previous training and experience in sampling for stable isotope analysis and a sound knowledge of the underlying theory is preferable. Some suggested reading is provided in section 8.

17.6. Equipment

Equipment and materials specific to this sampling method are included in (but not limited to) Appendix 1. Additional equipment may be needed depending on sample type (e.g. whether sampling vegetation, phytoplankton, animals or sediments).

17.7. Procedure

17.7.1. Sample collection

Obtaining a clean sample at the time of collection can save a substantial amount of SIA processing time in the laboratory. Aim to collect a clean sample of the material of interest by excluding (or picking out) potential contaminating material. If possible, rinse the sample when it is collected. All sample materials collected should be placed into clean, clearly labelled bags (e.g. labelled with site, sample type, sample date and replicate number).

17.7.1.1. Vegetation sampling

SIA can be carried out on terrestrial, semi-aquatic and aquatic plants. Samples are frozen after collection. For terrestrial vegetation (trees, shrubs, grasses etc.) leaves are typically sampled.

Different types of aquatic plants and algae may be sampled and processed for SIA including:

- floating macrophytes
- rooted submerged aquatic macrophytes
- filamentous algae and other attached visible algal material (on snags, rocks, mats of algae on sediments).

After collection, remove any particulate organic matter (POM) and sediment from the sample prior to freezing. This can be done in a number of ways:

- float the material off the sample
- hand pick the sample
- sieve the sample

Epiphytes are also commonly collected for SIA. Epiphytes are small plants without roots that grow on other plants. If they are available at the time of sampling, the best method of collection is by scraping them from the plant leaf using a scalpel, rinse and store in clean clip-seal bags prior to freezing.

All samples must be labelled correctly with details on the site where the samples were collected, time, date and sampler and given a unique identifier. In addition, record the type of sample collected, details on the part of the plant, preparation undertaken, preservation and sampler name.

17.7.1.2. Phytoplankton sampling

Water samples should be collected into clean containers and processed as soon as possible. The amount of water required depends on the chlorophyll content of the water. Sample collection and processing should be undertaken as outlined in the Chlorophyll a sample collection methods document. Glass fibre filters used in the collection of chlorophyll a samples for SIA should be pre-combusted and pre-weighed before use. Zooplankton and large detritus can be removed from the sample by pre-filtering water through a 100-200 μ m mesh sieve, or by removing large pieces with forceps.

All samples must be labelled correctly with details on the site where the samples were collected, time, date and sampler and given a unique identifier. This information must be recorded. In addition, record the type of sample collected, preparation undertaken, preservation and sampler name in a notebook or equivalent.

17.7.1.3. Animal sampling

Animals can be rinsed and frozen whole for processing later. If the animal is of substantial size (e.g. a large fish), measure standard length (**Figure 32**) and total weight of the fish before removing any tissue samples. When collecting samples from large fish for SIA:

- collect muscle (flesh) samples from the fish at the time of collection rather than transporting the entire animal
- collect any other samples such as gut contents, otoliths (for diet and aging purposes), or the liver (for shorter term diet indication) that are required
- bag and label each tissue sample separately for each individual fish.

Very small fish can be sent to the laboratory whole. It is recommended a number of small fish from a sampling site are sent to the laboratory so samples can be pooled if necessary.

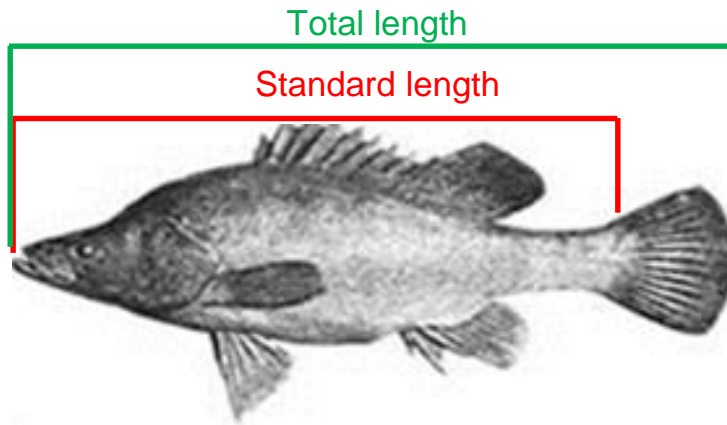


Figure 32: Fish schematic, showing total and standard length measurements

Some smaller animals such as worms and nematodes need to have the contents of their guts emptied.

- A. Place animals in filtered clean water (freshwater or seawater) in a labelled watertight plastic bag.
- B. Let sit for between 12 and 24 hrs to evacuate gut.
- C. Rinse samples in de-ionised water.
- D. Freeze samples as soon as possible

If this is not possible, rinse and freeze the animals; their guts can be removed later.

All samples must be labelled correctly with details on the site where the samples were collected, time, date and sampler and given a unique identifier. This information must be recorded. In addition, record samples collected, preparation undertaken, preservation and sampler name in a notebook or equivalent.

17.7.1.4. Sediment sampling

Sediment sampling should be used when sampling for sediment sources and deposited sediment for SIA. Sediments should be collected as outlined in the Collection and preservation of sediment document. Sediment samples can be size fractionated in the laboratory into different particle sizes for isotopic analysis of each size fraction if required. Selected size fractions for analysis would be defined by the aim of the study.

If suspended sediment is being studied, sampling equipment such as time integrated samplers (Phillips et al. 2000) or auto-samplers (see Water quality sampling using automated sampling equipment document) can be used.

For SIA sediment collection:

- Large pieces of organic matter, such as leaves and twigs, should be removed in the field with sieves or forceps.
- Coarse particulate organic matter (CPOM) >1.0mm and/or fine particulate organic matter (FPOM) <1.0mm should be collected using a series of sieves, as each size fraction contains different organic detritus which will be targeted during SIA analysis.
- Collected sampled soil and/or sediment should be bagged into clean snap-lock bags, labelled and put on ice.

Because of the high variability of C and N content in sediment samples, repeat/replicate analyses may be required. It is important therefore to collect and provide as much sample as possible for subsequent runs if required. If composite samples are collected, ensure that they are completely homogenised before sending to the laboratory.

All samples must be labelled correctly with details on the site where the samples were collected, time, date and sampler and given a unique identifier. This information must be recorded. In addition, record samples collected, preparation undertaken, preservation and sampler name in a notebook or equivalent.

17.7.2. Storage

After collection, all samples must be frozen immediately (to -18°C or lower) or stored on ice (do not use dry ice) until they can be frozen. Samples must remain frozen until processed. Chemical preservation and/or incorrect storage of samples (see below) may affect isotopic composition (e.g. Kaehler and Pakhomov 2001; Arrington and Winemiller 2002; Sarakinos et al. 2002).

Note: Preservatives **must not** be used on samples undergoing SIA.

17.7.3. Quality assurance and quality control

All storage containers must be clean, air tight and clearly labelled. Cross-contamination of samples must be avoided by using a clean set of processing equipment for each sample. Chain of custody documentation and clear and accurate sample data sheets must be provided to the laboratory.

17.7.4. Sample submission

Sample submission details should be discussed directly with the laboratory.

17.7.5. Variation to method

Different laboratories will have different requirements and the operating technician should be contacted prior to project commencement to determine any specific needs. Variations may include measurement of other isotopes such as oxygen, hydrogen or strontium and the inclusion of water samples for isotope analysis.

17.8. References and additional reading

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Appendix 1**Table 1: Equipment checklist**

Equipment	✓
A note book or equivalent for recording data.	
Distilled water	
Sample submission sheets	
Storage containers and bags	
Permanent, water proof marker	
Sieves	
Latex gloves (powder free)	
Aluminium pie dishes	
Analytical balance	

Section C: Biological Assessment

Background to aquatic macroinvertebrates sampling and index calculation

Version: February 2018

1. Background to aquatic macroinvertebrates sampling and index calculation

1.1. Purpose and Scope

This document and its companion document Aquatic macroinvertebrate sampling, processing and index calculation outline the methods used to derive the indices on which the aquatic macroinvertebrates water quality objectives (WQOs) (Environmental Protection (Water) Policy 2009) are based¹². When assessing whether a site meets the aquatic macroinvertebrate water quality objectives (WQOs) currently prescribed under the Environmental Protection (Water) Policy (2009), the live picking method must be followed, as this method was used to derive the published WQOs. Local WQOs may be derived using the laboratory picked method, provided adequate reference site data are available (in terms of number of sites and over time) (see EHP 2009 for guidance). The method used to define any WQOs should be clearly stated, and then consistently used for any comparisons.

1.2. Associated documents

Biological assessment: Aquatic macroinvertebrate sampling, processing and index calculation

1.3. Introduction

The structure of plant and animal communities of rivers can provide valuable information on the condition or health of waterways (in addition to water quality parameters). Aquatic macroinvertebrates are the most widely used biological indicators globally, because they are abundant and diverse, and can be sensitive to changes in water quality, flow regime and habitat conditions. Aquatic macroinvertebrates are animals without backbones and are large enough to be seen with the naked eye (e.g. prawns, shrimps, crayfish, snails, mussels and insects such as dragonflies, damselflies and mayflies).

Nationally, two methods are commonly used for collecting organisms: sampling a defined length of habitat using a dip net and either field picking or laboratory picking the sample. For Queensland, the field picking option has been used to derive aquatic macroinvertebrate WQOs that are defined for a number of river basins under the Environmental Protection (Water) Policy (2009). Locally relevant WQOs can be derived for either method, providing data are collected from enough reference sites over enough time (see EHP 2009 for guidance).

Macroinvertebrate indices, including those used to derive WQOs, can be insensitive to environmental change (including impacts from point source pollution). Therefore, it is recommended specialist advice be obtained when a study is being undertaken for any purpose other than comparison to WQOs. The study should be designed to fully address the complexity of the environmental problem being assessed. For example, the use of replicated quantitative macroinvertebrate sampling may be appropriate for assessing point source pollution (e.g. SSD 2013). Any environmental data (including macroinvertebrate data) used to assess environmental harm should be used as part of a multiple lines of evidence approach—a sole indicator of aquatic ecosystem impacts should not be used.

1.4. Aquatic habitats

A reach of a stream may have several habitats, each of which may have a different taxonomic composition. Bed samples can consist of riffles, pools, runs and macrophyte habitat. Riffles have a rocky bed (**Figure 33**), whereas runs (**Figure 34**) and pools (**Figure 35**) can consist of either rocky or sandy beds. Composite samples are a combination of all bed habitats present at a site.

The habitats most likely to be encountered in Queensland are summarised below. The definitions of pool, riffle and run can be confirmed by measuring and calculating the ratio of velocity (V) to depth (D). The definitions are based

¹² Further information can be found at <http://www.ehp.qld.gov.au/water/policy/>

on the following ratios: $V:D > 0.032$ = riffle; $V:D < 0.0124$ = pool; $V:D 0.0124-0.032$ = run.

A riffle (**Figure 33**) is a reach of relatively steep, shallow (generally $< 0.3\text{m}$), fast flowing ($\geq 0.2\text{m/s}$) and broken water over stony beds. A run (**Figure 34**) is a reach of flowing, unbroken water over a sandy, stony or rocky bed. Pool habitats (**Figure 35**) are relatively deep, stationary or very slow flowing water over silty, sandy, stony or rocky beds in the main channel. Edges (banks and under bank areas) (**Figure 36**) are habitats along the bank with little or no current. There may be some terrestrial vegetation (e.g. paragrass and sedges), tree roots or the area might be bare. A backwater is a zone where the bank indents and a pool of water forms away from the main channel (e.g. ox-bow, off-cut channel). It might have a circular or back flow, and a silty bed with accumulated plant litter. Macrophyte habitats (**Figure 37**) are areas where emergent, submergent and floating aquatic plants are present and can occur in still to fast flowing waters.

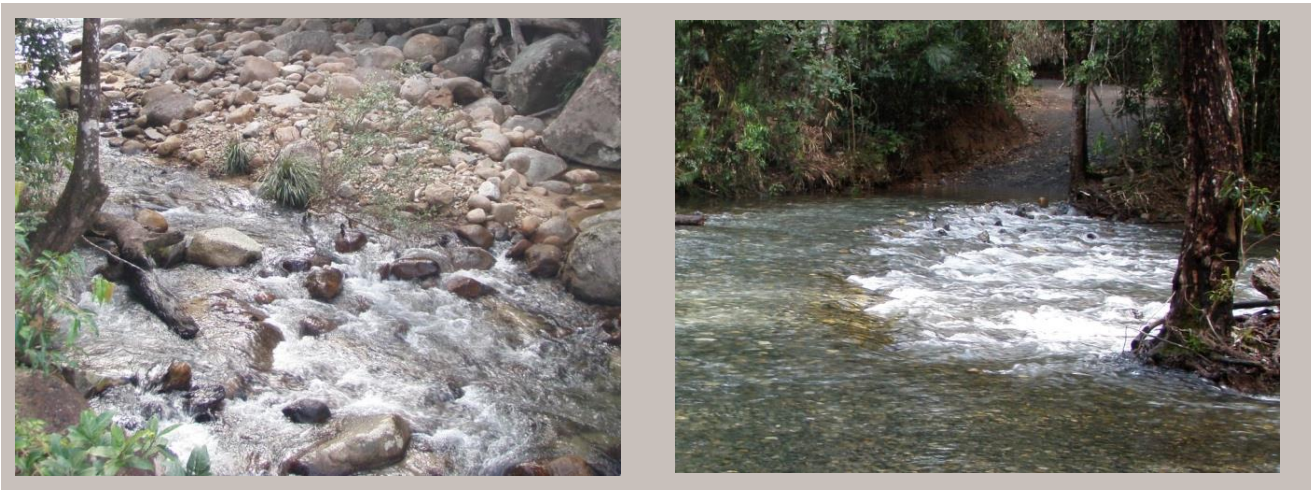


Figure 33: Riffle habitat

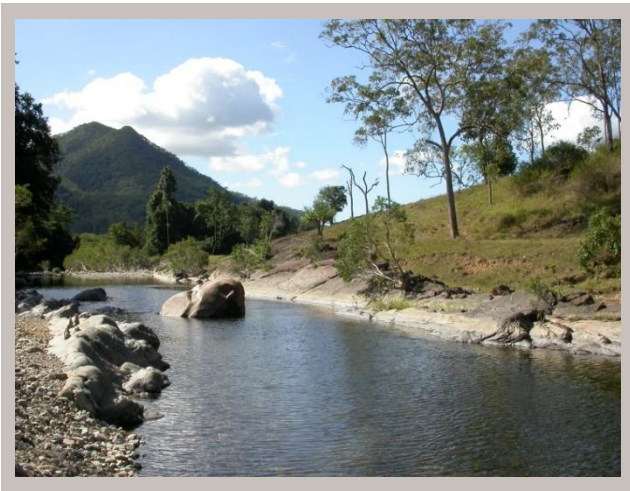


Figure 34: Example of run habitat



Figure 35: Pool habitats

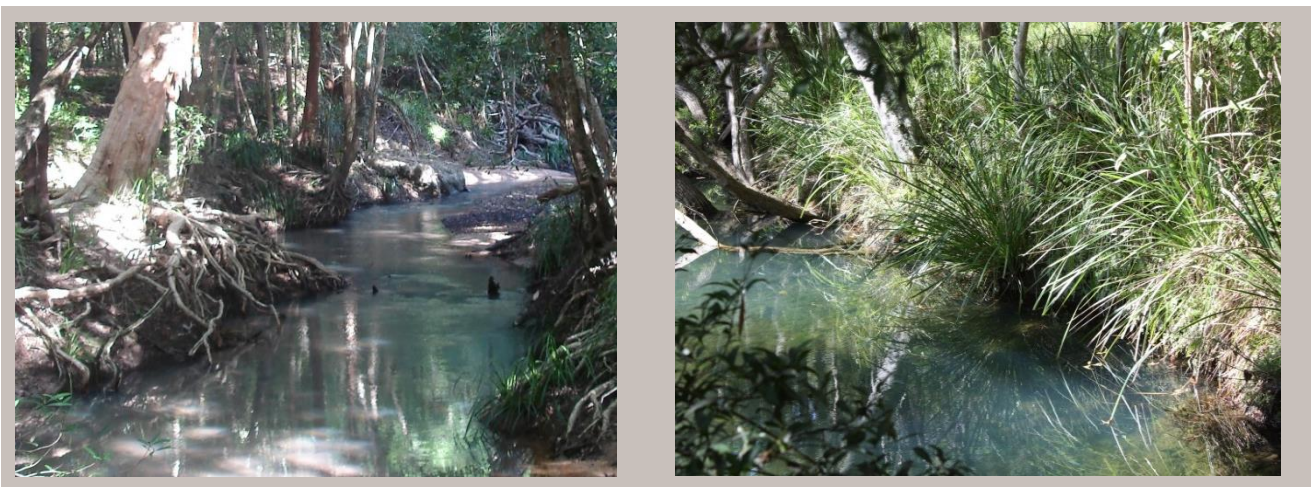


Figure 36: Edge habitat



Figure 37: Macrophyte habitat

1.5. Macroinvertebrate indices

Macroinvertebrate indices are used as a measure of ecosystem health. Five macroinvertebrate indices have been used to derive macroinvertebrate water quality objectives (WQOs) in Queensland, and are described below. A change from a WQO can indicate an impact from changed environmental conditions. This change can include an increase or a decrease in the values of the expected WQO. For example, nutrient and flow increases can increase richness. Increased conductivity can decrease all indices.

1.5.1. Richness

Richness is a count of the number of different macroinvertebrate taxa present at a site. WQOs for richness are given either as a single number or a range. If the taxa richness falls between the provided reference range or is close to the reference WQO (in the case of a single value) then the waterway is considered to be more likely in a good condition.

1.5.2. SIGNAL Index

The SIGNAL Index (Stream Invertebrate Grade Number - Average Level) was developed for the bioassessment of water quality in Australia (see Chessman 1995; 2001; 2003). A sensitivity grade number is allocated to different macroinvertebrate taxon/taxa. These sensitivity grades are based on how sensitive each taxon is to various pollutants. Grade numbers are between 1 and 10 and are available for most macroinvertebrate orders/families encountered in Queensland streams. The higher the SIGNAL value, the better the condition of the water quality at a site. SIGNAL is calculated as the arithmetic mean of the grade of each taxon within a sample.

1.5.3. PET taxa richness

Macroinvertebrates belonging to the PET (EPT) orders - Plecoptera, Ephemeroptera, and Trichoptera are considered to be particularly sensitive to changes in their environment (Karr and Chu 1999). Therefore PET taxa richness can be used to assess degradation of habitat and water quality (Plafkin et al. 1989; Barbour et al. 1992). The PET index is not necessarily a useful indicator for many Queensland rivers and streams as: Plecopterans are usually rare in Queensland rivers; Trichopterans and Ephemeropterans in Queensland can be tolerant to a range of conditions and may not be sensitive to many impacts; and some areas naturally don't contain these taxa. PET is a count of PET taxa.

1.5.4. Per cent sensitive taxa index

The per cent sensitive taxa index was developed to overcome the dominance of tolerant taxa in the calculation of the SIGNAL Index (which results in low variability in SIGNAL Index scores between sites in Queensland). If a site is experiencing an impact, it is expected that there would be a change in the percentage of sensitive taxa collected. For this index, taxa with SIGNAL grade numbers of 8 or greater are designated as sensitive (Marshall et al. 2001), and the number of these sensitive taxa are compared to the overall taxa count and expressed as a percentage.

1.5.5. Per cent tolerant taxa index

If a site is experiencing an impact it is expected that there would be a change in the percentage of tolerant taxa collected. The per cent tolerant taxa index is calculated from SIGNAL grade numbers. Taxa with SIGNAL grade numbers of 3 or less are designated as tolerant (Marshall et al. 2001), and the number of these tolerant taxa are compared to the overall taxa count and expressed as a percentage.

1.6. References and additional reading

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Aquatic macroinvertebrate sampling, processing and index calculation

Version: May 2018

2. Aquatic macroinvertebrate sampling, processing and index calculation

2.1. Purpose and scope

This document describes methods for aquatic macroinvertebrate sampling. Methods for both live picking and laboratory picking are presented. However, when assessing whether a site meets the aquatic macroinvertebrate water quality objectives (WQOs) prescribed under the Environmental Protection (Water) Policy (2009), the live picking method must be followed, as this method was used to derive the current published WQOs. Local WQOs may be derived using the laboratory picked method, provided adequate reference site data are available (in terms of number of sites and over time) (see EHP 2009 for guidance). The method used to define any WQOs should be clearly stated.

2.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological assessment: Background to aquatic macroinvertebrate sampling and index calculation.

2.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

2.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See *Permits and approvals* for more information on requirements.

Note: Crustaceans (e.g. crabs, prawns, crayfish and lobsters) are considered fish under the *Fisheries Act 1994* and therefore relevant permits and approvals need to be sought.

2.5. Skills, training and experience

There are four components in this method: sample collection, live picking, laboratory picking, and laboratory taxonomic identification. Skills, training and/or experience required to understand and/or undertake each of these components are:

- **Aquatic macroinvertebrate sample collection:** In house training by an accredited or equivalently trained operator at a range of habitats and sites.
- **Field (live) picking:** Completion of an aquatic macroinvertebrate ecology course and/or equivalent AUSRIVAS accreditation in **live** picking. In house training may be provided by an accredited or equivalently trained operator. The QA/QC procedure outlined in this document can be used to assess competency.
- **Laboratory picking:** Completion of an aquatic macroinvertebrate ecology course and/or equivalent AUSRIVAS accreditation in **laboratory** picking. In house training by an accredited or equivalently trained operator at a range of habitats and sites.
- **Aquatic macroinvertebrate identification:** Completion of an aquatic macroinvertebrate identification course and/or equivalent AUSRIVAS accreditation in aquatic macroinvertebrate identification. In house training may be provided by an accredited or equivalently trained operator. The QA/QC procedure outlined in this document can be used to assess competency.

2.6. Equipment

See Appendix 1 for example equipment checklist.

2.7. Procedure

Nationally, two methods are used for collecting organisms: field picking and laboratory picking. The choice of which method to adopt will be influenced by considerations of the objective of the study, precision required, time, cost and balance of effort in the field versus laboratory. Either method may be used, although it is critical to maintain the same technique for all sites when comparing data.

For Queensland, the field picking option has been adopted as the preferred standard method. This is the method that was used to collect data for the development of macroinvertebrate water quality objectives (WQOs) that are defined for a number of river basins under the Environmental Protection (Water) Policy (2009) (EPP (Water))

Note: If the stream being considered is experiencing high flow, or has recently filled after being dry, macroinvertebrate sampling should not be conducted. Once the high flow has subsided, a period of 4–6 weeks should be left before conducting sampling in order for hydrological conditions to stabilise and aquatic macroinvertebrate populations to recover.

2.7.1. Habitat selection

2.7.1.1. Sampling site and aquatic habitat

The site is defined as 50m upstream and 50m downstream of the entry point (total of 100m of the stream reach). Aquatic habitats are defined using a combination of visual identification and velocity to depth ratio validation. The key visual identifiers for each habitat type are summarised in Table 7. The velocity to depth table (Appendix 2) can be used to confirm the identification of a habitat type based on flow. Velocity should be calculated using a flow meter at several points along a stream cross section. Alternatively a visual estimator can be used, where the time it takes for an object to move down a known length of stream is measured. Depth can be measured using a depth measuring pole.

Table 7: Key descriptors of different habitat types

Habitat type	Key descriptor	Depth	Flow
Edge	Habitats along bank, approximately 0.5m from bank. May be terrestrial or semi-aquatic vegetation (e.g. paragrass and sedges), tree roots or bare area.		Little or no current
Backwater	Pool of water away from the main channel (e.g. ox-bow, off-cut channel). May have a silty bed with accumulated plant litter (e.g. leaves, twigs etc.)		Circular or back flow
Riffle (bed)	Broken water over stony beds	Shallow (<0.3m)	Fast ($\geq 0.2\text{m/s}$)
Run (bed)	Unbroken water	Deep	Fast flowing
Pool (bed)	Unbroken, main channel	Relatively deep	Stationary/slow
Macrophyte (bed)	Areas where emergent, submergent and floating macrophytes or aquatic plants are present	Relatively shallow	Slow or fast

Habitats to be sampled in each region to align with the EPP (Water) WQOs for macroinvertebrates are summarised in Table 8.

Table 8: Habitats to be sampled in each region to align with the EPP (Water) Water Quality Objectives (WQOs)

Areas with aquatic macroinvertebrate water quality objectives (WQOs) ¹³	Habitats assessed
Wet Tropics ^a	Edge and riffle
Fitzroy Basin waters	Edge and composite of all bed habitats
Mackay-Whitsundays ^b	Edge and riffle
Capricorn-Curtis Coast region ^c	Edge
Moreton Bay/south-east Queensland	Edge

except ^a Barron River Basin, ^b Burrum, Gregory, Isis, Cherwell and Elliott rivers, and ^c Curtis Island, Calliope River and Boyne River basins where WQOs are not available for aquatic macroinvertebrates.

2.7.2. Sampling aquatic macroinvertebrates using a dip net

2.7.2.1. Preparation

1. Check sampling area is of a wading depth i.e. maximum depth of 1–1.5m. For Workplace Health and Safety reasons, ensure it doesn't have a strong current and isn't slippery.
2. Check nets for damage or holes. Only use nets that are free of damage or holes.
3. Wash nets and buckets prior to sampling each site to remove animals left from previous sampling.
4. Record site information (for example site location, site code, date, time etc.).
5. Record GPS co-ordinate of site.

2.7.2.2. Sampling habitats

1. Sample 10m of each of the chosen habitats using the techniques described below. The 10m sample for each habitat type can be fragmented within the 100m reach of the site.
2. Within each habitat, there are usually several different microhabitats present (e.g. an edge habitat may have tree roots, paragrass, bare banks, or leaf litter). As many of these microhabitats as possible should be sampled.

2.7.2.2.1. Edge/backwater

1. Locate an edge area with little or no aquatic vegetation (stands of paragrass, *Urochloa* sp., are acceptable as edge habitat). An alcove or backwater with abundant benthic leaf litter is also acceptable. Suitable areas include tree roots, fine organic/silt deposits, and trailing vegetation, and are often indicated by the presence of surface-dwelling insects. Bare edges should also be sampled.
2. Working upstream when possible, use short forward and upward sweeping movements at right angles and towards the bank and continue sampling until the desired edge length/distance is reached. Stir up the benthos while doing so, ensuring that benthic animals are suspended and then caught when sweeping through the cloud of suspended material.
3. There may be aquatic plants along the banks and in backwaters. Avoid sampling these areas as part of an edge sample.

Note: Avoid clogging the net with fine particulate material. If necessary, flush the net with water without losing macroinvertebrates already captured.

2.7.2.2.2. Riffle/runs

1. While holding the net with its mouth facing upstream, disturb the substratum with the feet so that macroinvertebrates are washed into the net. Turn and rub cobbles and rocks by hand to dislodge organisms.
2. Continue this process walking backwards and working upstream, covering both the fastest and slowest flowing

¹³ Schedule 1 of the Environmental Protection (Water) Policy 2009

sections of the riffle/runs until a distance of 10m is sampled.

- Place several cobbles into the net and then transfer them into a bucket for direct picking of animals from the cobbles.

Note: Do not include material from macrophytes and/or wood debris located in the riffle.

2.7.2.2.3. Pools

- Disturb the substratum by kicking with your feet.
 - If the stream is flowing, hold the net downstream of feet with the mouth facing upstream into the area being disturbed.
 - If there is no flow use a short sweeping action with the net to stir up the bed.
- Continue this process walking backwards and working upstream over the required distance.
- If the bed is rocky and the rocks are too large to kick over, in addition to sweeping over a distance of 10m, remove about 10 rocks of a range of sizes and wash the macroinvertebrates from them into the net by scrubbing gently with the hands or a light brush. Leave the rocks out of the water to allow cryptic specimens to emerge. These can then be hand-picked, using tweezers. Leaving the rocks in the sun for too long will dry out and kill the animals.

Note: Both Runs and Pools can have silty/sandy and rocky/gravel beds. When sampling silty/sandy beds, select an area with plant litter or periphyton (not macrophytes) rather than an area of clean sand.

2.7.2.2.4. Composite bed

- Sample 5m of each available bed habitat at the site, one after the other into the one net, using the methods described in Section 2.7.2.2.2 and 2.7.2.2.3.

Note: If there is only one type of bed habitat present at the site, sample 10m of this habitat.

2.7.2.3. Completion of sampling

2.7.2.3.1. For live picking of sample

- After sampling a habitat, empty the contents of the net into a bucket by inverting the net.
- Splash water onto the inverted net to wash any remaining animals into the bucket.

2.7.2.3.2. For laboratory picking of sample

- Rinse net sides to consolidate sample at the bottom of the net.
- Place a medium sized tray on the ground, and turn the net inside out so the sample falls into the tray.
- Place material collected in the tray into a sample container. Large plastic screw-top jars or heavy-duty plastic bags stored in a polydrum are suitable containers.
- Rinse the net into the tray to remove any remaining material.
- Tip water from tray through a 250µm sieved to collect remaining material. Rinse tray if necessary to remove all material. Place material collected in the sieve into the sample container.
- Using forceps, check the net for any remaining invertebrates and place into sample container
- Ensure a completed label is placed in the vial noting the project name, site number and name, sampling date, habitat sampled, sample collector and picker and any relevant notes (**Figure 38**). Label must be completed using pencil or alcohol-proof ink and should be on waterproof paper. Labels should also be printed in alcohol-proof ink.
- If more than one container is required to store the sample, clearly indicate this when labelling the containers, e.g., if two containers are required, label the containers 1 of 2 and 2 of 2.
- Preserve the sample with 70 % alcohol.

Project Name: _____	Date: ____ / ____ / ____
Site Name: _____	
Site No: _____	Habitat: _____
Collector: _____	
Collecting Method: _____	
Picking Method: _____	

Figure 38: Label for laboratory picked sample

2.7.3. Live picking aquatic macroinvertebrates

2.7.3.1. Sample preparation

If the sample has a large amount of detrital material, it is advisable to separate it into two using a 1cm panning sieve. Place each fraction in a separate bucket or tray; making sure that there is water in the bucket to prevent animals from desiccating.

Note: Samples should be picked as soon as possible after collection. Keep samples wet, cool and tightly covered if there is any delay in picking. A single operator picks the sample.

2.7.3.2. Method

- Set up table and chairs with timer, white trays, picking tool and vial or jar half filled with 70% ethanol or methylated spirits. For one of the first three samples collected in a sampling program, the procedure in Section 2.7.3.4 must be followed.
- Place a small amount of the sample material and water into a white tray.
- Start the timer.
- Pick aquatic macroinvertebrates from the tray using tweezers, spoon and pipettes, and transfer into the vial with the ethanol (or methylated spirits). Suck up small taxa, e.g. mites, with a pipette.
 - Aim to collect only 10 individuals of any one taxon, i.e. do not spend a large proportion of picking time concentrating on animals that all appear to be the same. When uncertain of the identity, then collect all of the organisms. At least 30 midge larvae (Chironomidae) should be collected to ensure adequate representation of the sub-families. The aim of live-picking is essentially to collect the greatest number of taxa as possible from the sample. Ten individuals of apparent taxa provides some indication of abundance and assists in collecting taxa that are visually similar.
 - At the start of the live pick, the common and abundant taxa should be targeted for the first 5 minutes. After that, the major picking effort should be directed at finding the less common, inconspicuous taxa.
- Work progressively through the sample, replacing picked material with fresh sample.
- After picking for 30 minutes, search specifically for new taxa for 10 minutes. If any new taxa are found in these 10 minutes, extend the picking time by another 10 minutes.
- Follow this procedure until either no new taxa are found or a maximum of 60 minutes have been spent on picking.
- Record the picking time, and add to the vial label (**Figure 39**).
- Record if the sample has very few animals and any likely reasons why that may be so (e.g. very silty/sandy bed). Add note to vial label (**Figure 39**).

Note:

- Ensure that you search through all components of the sample, including the sediment at the bottom of the bucket. Very occasionally, the collected sample is so large that it cannot be sorted adequately in the given time. In these circumstances, ensure that you have picked over all the different types of detritus, substrate and water-borne material present in your sample in the 60 minutes.
- If it is raining, cold or extreme heat, or conditions of poor light exist due to cloud cover or approaching twilight, the sample must be taken back to the vehicle/motel/camp etc. for sorting under cover and with improved light conditions. It is best to avoid these situations in the first place.

Live Picked Macroinvertebrate Sample	
Project:	<u>North QLD Bioassessment Program</u>
Site code:	<u>NQBPO01</u>
Site name:	<u>Tully River @ Brosnan Rd</u>
Date:	<u>22 / 03 / 2015</u> Time: <u>11:45</u> <u>am</u> /pm
Habitat sampled:	<u>Edge</u> Preservative: <u>Ethanol</u>
Collector:	<u>SKL</u> Picker: <u>GTH</u> Vial: <u>1</u> of <u>1</u>
Collection method:	<u>10m sweep</u>
Picking time/method:	<u>40min live pick</u>

Figure 39: Example of a vial label for aquatic macroinvertebrate samples picked in the field

2.7.3.3. Curation and storage

1. When picking is completed, remove some of the liquid in the vial (which is usually diluted by water that has been added with the animals) using a mesh-covered syringe (**Figure 40**).
2. Refill the vial with fresh 70% ethanol or methylated spirits to the top and ensure the lid is tightly screwed on.
3. Ensure the vial you use is large enough. If the animals you collect take up more than 30% of the volume, use a larger container. Alternatively, use several vials clearly labelled to indicate that more than one vial was used e.g. 'vial 1 of 3'.
4. A label must be placed in the vial, an example of the kind of information that should accompany the sample is presented in **Figure 39**. Label must be completed using pencil or alcohol-proof ink. Labels should also be printed in alcohol-proof ink.
5. Samples should be packed upright, in rigid lidded containers. Foam inserts are useful to ensure samples remain upright.



Figure 40: Mesh covered syringe

2.7.3.4. Quality assurance and quality control of live-picked aquatic macroinvertebrates

2.7.3.4.1. Field-based review of live picking

For one of the first three live picks of a sampling round, the following procedure must be followed:

1. The sample is live picked by the operator (Section 2.7.3.2). During the pick, a field aquatic macroinvertebrate record sheet is filled in (Appendix 3).
2. An immediate re-pick is then undertaken by a second operator on the residue of the sample picked by the first operator. The second operator must be trained and accredited. The re-pick is undertaken in the same way as a normal field live-pick and the field aquatic macroinvertebrate record sheet completed by the first operator is also filled in by the second operator.
3. The field aquatic macroinvertebrate record sheet is immediately checked for inconsistencies, which are discussed between the two pickers to reduce systematic errors in subsequent live picks. If errors are large a check of a second sample should be considered.

- Both samples are then preserved and transported to the laboratory for identification and further analysis.

2.7.3.4.2. Laboratory assessment of live pick

- Samples from both operators are processed as outlined in Section 2.7.4.
- Two statistical measures are then applied to the data:
 - The ratio of the live pick taxa richness from operator 1 to the total QA/QC sample taxa richness (sample from operator 1 + sample from operator 2) must be >0.90 .
 - The Bray-Curtis Dissimilarity Index between samples picked by operator 1 and the total QA/QC sample taxa richness (sample from operator 1 + sample from operator 2) must be <0.2 . Bray-Curtis Dissimilarity Index is based on presence/absence data.

Failure to meet these standards triggers a review of the operator's skills including consideration of re-training as required. Results of the quality assurance and quality control program should be presented in any report as evidence of quality in the results.

2.7.4. Laboratory picking aquatic macroinvertebrates

- Tip the preserved sample into a series of 10mm and 250mm sieves and thoroughly wash the sample.
- If there are large coarse fractions (sticks, leaves, etc.) wash these over the sieves and place them into a sorting tray. Examine these coarse fractions preferably using a magnifying glass, for approximately 10 minutes, ensuring that any macroinvertebrates attached to the coarse fractions are collected.

Note: keep an eye out for stick and leaf-cased Trichoptera.

- Evenly distribute the remaining smaller fractions from the sieves into a subsampler. The subsampler used and recommended is a modified Marchant subsampler (Marchant, 1989) (**Figure 41**). This subsampler contains 100 circular cells, each 3.5cm in diameter x 3.5cm deep.
- Fill the subsampler until the water level reaches the top of the cells, secure the lid, and rotate vigorously in both directions until the sample is distributed throughout the cells. Using a vacuum pump, subsample 10% (10 cells) of the whole sample, ensuring every 1% (1 cell) of the subsample is stored in separate containers. Label each container.
- Sort and identify the subsamples (see sections 2.7.5.2 and 2.7.5.3 for a detailed method details), noting how many new taxa there are in every 1% subsample sorted.

Note: Other types of subsamplers may be used if a Marchant subsampler is not available. Overall, 10 % of the sample should be subsampled.

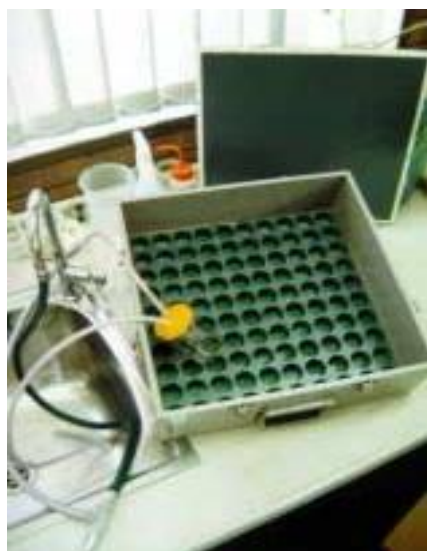


Figure 41: Modified Marchant subsampler

2.7.5. Identification and enumeration of aquatic macroinvertebrates to family level

2.7.5.1. Sample preparation

- Rinse the preservative from the sample by placing it in a 250µm sieve and gently running water through it, or

use a plastic 250ml specimen jar with a mesh insert in the lid (**Figure 42**).

- Using a water squeeze bottle, flush the sieved contents into a large petri dish or sorting tray lid (**Figure 42**). If the sample is large flush the sieve into a 250mL specimen jar. Small amounts of the sample and water can then be poured into a sorting tray or petri dish for processing. If there is excess water in the sample it can be removed using a 250µm mesh-covered syringe.

Note: Always ensure that there is enough water left to completely cover all animals. When finished or if processing is interrupted for an extended period of time (e.g. overnight) replace the water with preservative (70% ethanol/methylated spirits and 3% glycerol).

2.7.5.2. Sample processing

- Place sorting tray under a dissecting microscope (minimum 10x magnification) and search systematically for aquatic macroinvertebrates.
- Remove the animals from the sorting tray under the lowest magnification and place in water in small petri dishes or glass specimen blocks for later identification. Use a separate dish for each type of taxa. It may be desirable to separate the adults, larvae, and pupae of some taxa such as beetles (adults and larvae) and dipterans (larvae and pupae), for ease of identification.
- Work your way through the sorting tray until all specimens have been removed.
- Adjust the microscope to a higher magnification and pass over the sorting tray once more. This will ensure that the smaller specimens that may be missed at a lower magnification are seen.
- Discard any non-macroinvertebrate material that may have been collected.
- Select a polypropylene vial or glass jar that is large enough to fit the entire sample and a label. Use pencil or alcohol-proof ink to fill in details. The label must have all the information presented in **Figure 43**.
- Place the label in the vial and half fill the vial with preservative (70% ethanol (or methylated spirits)/3% glycerol/27% water).
- Select a group of specimens and identify each specimen using the appropriate taxonomic keys. Organisms are identified to various taxa level as outlined in the example *Aquatic Macroinvertebrate Field Record Sheet* presented in Appendix 3. If uncertain about the identity, obtain a second opinion from a colleague or local specialist. If a new taxon is suspected, contact a taxonomic specialist or alternatively the Queensland Museum.
- When the specimen has been identified, place it in the vial and tally in the identification sheet (see example sheet provided in Appendix 4).
- Once all taxa have been identified and placed in the vial, fill it with preservative (70% ethanol (or methylated spirits)/3% glycerol/27% water) and screw the lid on firmly.
- Record the total tally for each taxon.

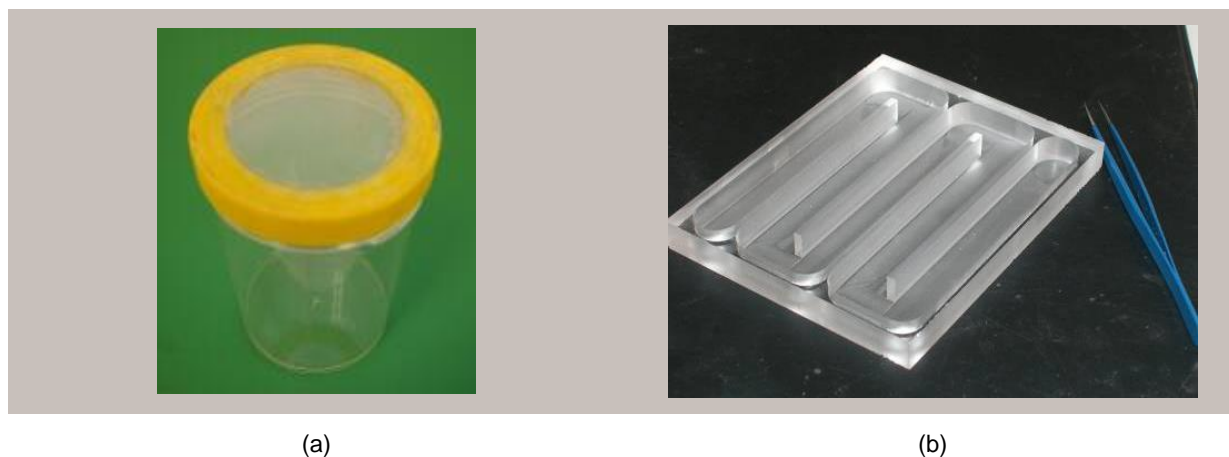


Figure 42: Example laboratory equipment used for aquatic macroinvertebrate sorting – (a) 250mL specimen jar with a mesh insert in the lid and (b) sorting tray and fine forceps.

Identified Macroinvertebrate Sample	
Project:	<u>North QLD Bioassessment Program</u>
Site code:	<u>NQB001</u>
Site name:	<u>Tully River @ Brosnan Rd</u>
Date:	<u>22 / 03 / 2015</u> Time: <u>11:45</u> <u>am</u> /pm
Habitat sampled:	<u>Edge</u> Taxa: <u>All</u>
Collector:	<u>SKL</u> Picker: <u>GTH</u>
ID by:	<u>PRT</u> ID date: <u>01 / 04 / 2015</u>
Vial	<u>1</u> of <u>1</u> Preservative: <u>70% Ethanol</u>

Figure 43: Example laboratory vial label

2.7.5.3. Quality assurance of taxonomic identification of aquatic macroinvertebrates to family level

2.7.5.3.1. QA/QC requirements

For each person undertaking aquatic macroinvertebrate identification (the operator) the following checks should occur:

1. The first five samples processed from a sampling round are to be re-checked by someone who is accredited (or equivalent) to identify aquatic macroinvertebrates.
2. Ten per cent of each operator's samples must be randomly selected for re-identification. These must not include the initial five samples.

For the samples that are undergoing a QA/QC check:

3. A QA/QC laboratory tally sheet (Appendix 5) is used to record the identities and numbers of all taxa in a sample. Two columns are provided so that the original tallies can be recorded alongside the checked tallies. Original tallies should only be obtained after the check has been completed.
4. If there are discrepancies in either identification these should be checked. This may entail checking that all animals were flushed from the vial and label, that all the animals were removed from the sorting tray to the holding dishes, and that there are no small animals hidden by or attached to larger ones.

2.7.5.3.2. Calculation of quality assurance results

Determining whether or not an operator meets laboratory QA/QC standards can be derived from two statistical measures:

- Ratio of taxon richness in the original sample identification (by the operator) and QA identification (by accredited person) (see section 7.5.3.1) must be >0.90
- Number of correct identifications (>90%).

Failure to meet these standards triggers a review of the operator's skills including consideration of re-training as required.

2.7.6. Long-term sample preservation and storage

1. Samples in small vials can be permanently stored in an evaporation-proof container such as a large 2L glass jar (polypropylene containers are also appropriate) filled with preservative to ensure individual samples do not dry-out. If a jar is under-filled, the remaining space should be filled with cotton wool to reduce movement of the vials in the jar.
2. The jar must be labelled with a unique number, with the project name, dates and sample identifiers (e.g. Samples 45-104) written on a label so it can be seen from the outside of the jar.
3. Details of the contents and location of each jar should be stored in a central location, ideally in an electronic format to enable samples to be located at a future date.
4. The jars must be checked annually and topped up with the preservative if required. A local schedule for maintenance should be implemented to ensure this occurs.

2.7.7. Calculation of richness, SIGNAL, PET taxa richness, per cent sensitive taxa and per cent tolerant taxa

2.7.7.1. Richness (taxa) index

Count the number of different taxa at the relevant level of taxonomic resolution (as outlined in the aquatic macroinvertebrate laboratory identification sheet – Appendix 4).

2.7.7.2. Average SIGNAL index calculation

1. Allocate a SIGNAL grade number to each taxon in the sample. SIGNAL grade numbers for version 2.4 are available from the SIGNAL manual (Chessman 2003) which can be found at <http://www.environment.gov.au/resource/signal-2iv-scoring-system-macroinvertebrates-water-bugs-australian-rivers>.
2. The SIGNAL Index is calculated for each sample by averaging the SIGNAL grade numbers of all of the aquatic macroinvertebrate taxa collected in a sample.
3. Taxa that do not have a SIGNAL grade number, for example Copepoda, Cladocera and Ostracoda, are not used in the calculation of the SIGNAL Index.

2.7.7.3. PET taxa richness

PET taxa richness is the count of families (or genera/species) that belong to the following three orders of aquatic macroinvertebrates: Plecoptera (stoneflies), Ephemeroptera (mayflies) and Trichoptera (caddisflies).

2.7.7.4. Calculating the % sensitive taxa index using presence/absence data

1. Count the number of taxa in a sample that have SIGNAL grade numbers.
2. Count the number of taxa that are sensitive (SIGNAL grade ≥ 8).
3. Calculate the per cent sensitive taxa using the formula below:

$$\% \text{ Sensitive Taxa Index} = \frac{\text{Number of taxa with SIGNAL grade numbers} \geq 8}{\text{Total number of taxa in sample with SIGNAL grade numbers}} \times 100$$

2.7.7.5. Calculating the % tolerant taxa index using presence/absence data

1. Count the total number of taxa in a sample that have SIGNAL grade numbers.
2. Count the number of taxa that are sensitive (SIGNAL grade ≤ 3).
3. Calculate the per cent sensitive taxa using the formula below:

$$\% \text{ Tolerant Taxa Index} = \frac{\text{Number of taxa with SIGNAL grade numbers} \leq 3}{\text{Total number of taxa in sample that have SIGNAL grade numbers}} \times 100$$

2.8. References and additional reading

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Appendix 1 Equipment checklist

Table 1 Aquatic habitat identification

Equipment	✓
Flow current meter	
Depth measuring device (ruler or pole with tape measure)	

Table 2 Sampling aquatic macroinvertebrates using a dip net

Equipment	✓
1 x net (Triangular 250mm x 250mm x 250mm opening, 1-1.5m handle, Mesh: 250µm with depth of 50-75cm). Spare net or equipment to mend net.	
3 x buckets	
Note book or field sampling sheet	
Pens, pencils, waterproof markers	
Waders	

Table 3 Live picking aquatic macroinvertebrates

Equipment	✓
Large white plastic trays (minimum 2) (approx. 40cm x 30cm x 6cm)	
Picking tools: very fine forceps, plastic Pasteur pipettes, plastic spoon	
Timer	
Buckets (minimum 2 per sample)	
Sieve (1cm mesh, 30cm diameter)	
Table and two chairs	
Alcohol stable 30mL, 70mL, 120mL and 250mL plastic screw top vials/jars	
Note book or field sampling sheet	
Alcohol stable sample labels (Figure 39)	
Mesh-covered syringe	
250µm mesh dip net, or 250µm mesh sieve	
Pencil or alcohol-proof ink	
Container with lid for picked sample vials preferable with foam insert	
70% ethanol or methylated spirits	

Table 4 Identification of aquatic macroinvertebrates

Equipment	✓
250µm sieve, or plastic 250ml specimen jar with a mesh insert in the lid	
Dissecting stereomicroscope (x6 to x50 magnification)	
Sorting tray or large glass petri dish	
Glass petri dishes/glass specimen blocks (watchglasses)	
Fine forceps	
Alcohol and water wash bottles	
Storage vials (polypropylene) 5ml, 25ml	
Glass storage jars (wide mouth) 70ml	
Vial and curatorial jars labels	
Laboratory identification sheet (Appendix 4)	
Taxonomic (identification) keys	
Pencil or alcohol-proof ink	
Cotton wool or cardboard filler, if required	
70% ethanol or methylated spirits	
Mesh covered syringe	

Appendix 2 Velocity: Depth table

For use in identifying aquatic habitats based on Velocity to Depth ratio.

$V:D > 0.032$ = riffle (R=Riffle in Table 5)

$V:D < 0.0124$ = pool (P=Pool in Table 5)

$V:D 0.0124 - 0.032$ = run (Run in Table 5)

Table 5 The Velocity:Depth table to identify stream habitat types (R = riffle and P = pool)

Depth (cm)	Velocity (m/sec)																									
	0	0.1	0.2	0.3	0.4	0.5	0.6	0.7	0.8	0.9	1.0	1.1	1.2	1.3	1.4	1.5	1.6	1.7	1.8	1.9	2.0	2.1	2.2	2.3	2.4	2.5
5	P	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
10	P	P	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
15	P	P	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
20	P	P	P	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
25	P	P	P	P	run	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
30	P	P	P	P	run	run	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
35	P	P	P	P	P	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R
40	P	P	P	P	P	run	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R
45	P	P	P	P	P	P	run	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R
50	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R	R
55	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R
60	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R	R
65	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R
70	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	R	R
75	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	R
80	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
85	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
90	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
95	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
100	P	P	P	P	P	P	P	P	P	P	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run

**Appendix 3 Example aquatic macroinvertebrate field record sheet
(appears on following page)**

Aquatic Macroinvertebrate Field Record Sheet

Project: _____	Vial: ___ of ___ (e.g. 2 of 3)	Collectors initials: ___ ___
Site code: _____	Habitat: _____ (e.g. Edge)	
Site name: _____	Picker 1 name: _____	Picker 2 name: _____
Sampling run: _____	Picker 1 (original picker) signature: _____	Picker 2 (re-picker) signature: _____
Sampling date: ___ / ___ / 20___		

Picker 1 (original picker): tally number of specimens in column "1" next to the correct taxonomic group

Picker 2 (re-picker): tally number of specimens in column "2" next to the correct taxonomic group

	1	2		1	2		1	2		1	2			
Lower Phyla			Glacidorbidae			s-f Tanypodinae			Georissidae			Zygotera		
Rotifera			Tateidae [ex-Hydrobiidae]			Culicidae			Gyrinidae			Argiolestidae [ex-Megapodagrionidae]		
Porifera			Lymnaeidae			Dixidae			Haliplidae			Calopterygidae		
Hydridae			Neritidae			Dolichopodidae			Heteroceridae			Chorismagrionidae		
Clavidae			Physidae			Empididae			Hydraenidae			Coenagrionidae		
Dugesidae			Planorbidae			Ephyridae			Hydrochidae			Diphlebiidae		
Temnocephaloidea			Thiaridae			Muscidae			Hydrophilidae			Isostictidae		
Nemertea			Viviparidae			Psychodidae			Hygrobidae			Lestidae		
Nematoda			Corbiculidae			Sciomyzidae			Limnichidae			Platycnemidae [ex-Protoneuridae]		
Gordiidae			Hyriidae			Simuliidae			Nanophyidae [ex-Brentidae]			Synlestidae		
Oligochaeta			Sphaeriidae			Stratiomyidae			Noteridae			Epiprocta		
Polychaeta			Megaloptera			Tabanidae			Psephenidae [adults terrestrial]			Aeshnidae		
Hirudinea			Corydalidae			Tanyderidae			Ptilodactylidae			Austrocorduliidae		
Glossiphoniidae			Sialidae			Thaumaleidae			Scirtidae			Cordulephyidae		
Hirudinidae			Ephemeroptera			Tipulidae			Spercheidae			Corduliidae		
Erpobdellidae			Ameletopsidae			Hemiptera			Sphaeriusidae [ex-Microsporidae]			Gomphidae		
Acarina			Baetidae			Aphelocheiridae			Staphylinidae			Hemicorduliidae		
Collembola			Caenidae			Belostomatidae			Trichoptera			Libellulidae		
Sminthuridae			Ephemerellidae			Corixidae			Antipodeciidae			Lindeniidae		
Crustacea			Leptophlebiidae			Gelastocoridae			Atriplectididae			Macromiidae		
Spinicaudata			Prosopistomatidae			Gerridae			Calamoceratidae			Pseudocorduliidae		
Cladocera			Neuroptera			Hebridae			Calocidae			Synthemistidae		
Ostracoda			Neurorthidae			Hydrometridae			Conoesucidae			Telephlebiidae		
Copepoda			Osmylidae			Leptopodidae			Dipseudopsidae			Urothemistidae		
Notostraca			Sisyridae			Mesoveliidae			Ecnomidae			Moths		
Psammaspididae			Plecoptera			Micronectidae			Glossosomatidae			Crambidae [ex-Pyralidae]		
Amphi-Chiltoniidae [Ex. Ceinidae]			Austroperlidae			Naucoridae			Helicophidae					
Amphi-Corophiidae			Eustheniidae			Nepidae			Helicopsychidae			OTHER TAXA		
Amphi-Eusiridae			Gripopterygidae			Notonectidae			Hydrobiosidae					
Iso-Sphaeromatidae			Notonemouridae			Ochteridae			Hydropsychidae					
Iso-Corallanidae			Diptera			Pleidae			Hydroptilidae					
Atyidae			Athericidae			Saldidae			Leptoceridae					
Palaemonidae			Blephaceridae			Veliidae			Odontoceridae					
Parastacidae			Ceratopogonidae			Coleoptera			Philopotamidae					
Parathelphusidae			Corethrellidae			Carabidae			Philorhreithridae					
Trogloplacidae			Chaoboridae			Chrysomelidae			Polycentropodidae					
Mollusca			Chironomidae			Curculionidae			Psychomyiidae					
Ancylidae [part Planorbidae]			s-f Chironominae			Dytiscidae			Tasimiidae					
Bithyniidae			s-f Orthoclaadiinae			Elmidae			Stenopsychidae					

**Appendix 4 Example of a laboratory identification sheet
(appears on following page)**

Aquatic Macroinvertebrate Identification Sheet

Project: _____		
Site code: _____	Vial: ___ of ___ (e.g. 2 of 3)	I.D. by: _____
Site name: _____	Habitat: _____ (e.g. Edge)	I.D. date: ___ / ___ / 20___
Sampling run: _____	Collectors initials: _____	Entered by: _____
Sampling date: ___ / ___ / 20___	Picker initials: _____	Entry checked by: _____

Counts by taxa

Lower Phyla	Glacidorbidae Tateidae [ex-Hydrobiidae] Lymnaeidae Neritidae Physidae Planorbidae Thiaridae Viviparidae Corbiculidae Hyriidae Sphaeriidae Megaloptera Corydalidae Sialidae Ephemeroptera Ameletopsidae Baetidae Caenidae Ephemerellidae Leptophlebiidae Prosopistomatidae Neuroptera Neurorthidae Osmylidae Sisyridae Plecoptera Austroperlidae Eustheniidae Gripopterygidae Notonemouridae Diptera Athericidae Blephariceridae Ceratopogonidae Corethrellidae Chaoboridae Chironomidae s-f Chironominae s-f Orthoclaadiinae	s-f Tanypodinae Culicidae Dixidae Dolichopodidae Empididae Ephyridae Muscidae Psychodidae Sciomyzidae Simuliidae Stratiomyidae Tabanidae Tanyderidae Thaumaleidae Tipulidae Hemiptera Aphelocheiridae Belostomatidae Corixidae Gelastocoridae Gerridae Hebridae Hydrometridae Leptopodidae Mesoveliidae Micronectidae Naucoridae Nepidae Notonectidae Ochteridae Pleidae Saldidae Veliidae Coleoptera Carabidae Chrysomelidae Curculionidae Dytiscidae Elmidae	Georissidae Gyrinidae Haliplidae Heteroceridae Hydraenidae Hydrochidae Hydrophilidae Hygrobiidae Limnichidae Nanophyidae [ex-Brentidae] Noteridae Psephenidae [adults terrestrial] Ptilodactylidae Scirtidae Spercheidae Sphaeriusidae [ex-Microsporidae] Staphylinidae Trichoptera Antipodeciidae Atriplectididae Calamoceratidae Calocidae Conoesucidae Dipseudopsidae Ecnomidae Glossosomatidae Helicophidae Helicopsychidae Hydrobiosidae Hydropsychidae Hydroptilidae Leptoceridae Odontoceridae Philopotamidae Philorhethridae Polycentropodidae Psychomyiidae Tasimiidae Stenopsychidae	Zygoptera Argiolestidae [ex- Megapodagrionidae] Calopterygidae Chorismagrionidae Coenagrionidae Diphlebiidae Isostictidae Lestidae Platycnemidae [ex-Protoneuridae] Synlestidae Epiprocta Aeshnidae Austrocorduliidae Ptilodactylidae Cordulephyidae Corduliidae Gomphidae Hemicorduliidae Libellulidae Lindeniidae Macromiidae Pseudocorduliidae Synthemistidae Telephlebiidae Urothemistidae Moths Crambidae [ex-Pyalidae] OTHER TAXA
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NOTES [e.g. taxa kept aside/sent away for expert ID]: _____

**Appendix 5 Example of a QA/QC laboratory identification sheet
(appears on following page)**

Aquatic Macroinvertebrate Identification Sheet – QA/QC									
Project: _____		Vial: ___ of ___ (e.g. 2 of 3)			QA/QC date: ___ / ___ / 20___				
Site code: _____		Habitat: _____ (e.g. Edge)			QA/QC I.D. by: ___ ___				
Site name: _____		Picker initials: ___ ___			Verification by: ___ ___				
Sampling run: _____		Original I.D. by: ___ ___			Entered by: ___ ___				
Sampling date: ___ / ___ / 20___		I.D. date: ___ / ___ / 20___			Entry checked by: ___ ___				
Counts by taxa	QA		QA		QA		QA		QA
Lower Phyla		Glacidorbidae		s-f Tanypodinae		Georissidae		Zygoptera	
Rotifera		Tateidae [ex-Hydrobiidae]		Culicidae		Gyrinidae		Argiolestidae [ex-Megapodagrionidae]	
Porifera		Lymnaeidae		Dixidae		Haliplidae		Calopterygidae	
Hydridae		Neritidae		Dolichopodidae		Heteroceridae		Chorismagrionidae	
Clavidae		Physidae		Empididae		Hydraenidae		Coenagrionidae	
Dugesiididae		Planorbidae		Ephydriidae		Hydrochidae		Diphlebiidae	
Temnocephaloidea		Thiaridae		Muscidae		Hydrophilidae		Isostictidae	
Nemertea		Viviparidae		Psychodidae		Hygrobiidae		Lestidae	
Nematoda		Corbiculidae		Sciomyzidae		Limnichidae		Platynemidae [ex-Protoneuridae]	
Gordiidae		Hyriidae		Simuliidae		Nanophyidae [ex-Brentidae]		Synlestidae	
Oligochaeta		Sphaeriidae		Stratiomyidae		Noteridae		Epiprocta	
Polychaeta		Megaloptera		Tabanidae		Psephenidae [adults terrestrial]		Aeshnidae	
Hirudinea		Corydalidae		Tanyderidae		Ptilodactylidae		Austrocorduliidae	
Glossiphoniidae		Sialidae		Thaumaleidae		Scirtidae		Cordulephyidae	
Hirudinidae		Ephemeroptera		Tipulidae		Spercheidae		Corduliidae	
Erpobdellidae		Ameletopsidae		Hemiptera		Sphaeriusidae [ex-Microsporidae]		Gomphidae	
Acarina		Baetidae		Aphelocheiridae		Staphylinidae		Hemicorduliidae	
Collembola		Caenidae		Belostomatidae		Trichoptera		Libellulidae	
Sminthurididae		Ephemerellidae		Corixidae		Antipodeciidae		Lindeniidae	
Crustacea		Leptophlebiidae		Gelastocoridae		Atriplectididae		Macromiidae	
Spinicaudata		Prosopistomatidae		Gerridae		Calamoceratidae		Pseudocorduliidae	
Cladocera		Neuroptera		Hebridae		Calocidae		Synthemistidae	
Ostracoda		Neurorthidae		Hydrometridae		Conoesucidae		Telephlebiidae	
Copepoda		Osmylidae		Leptopodidae		Dipseudopsidae		Urothemistidae	
Notostraca		Sisyridae		Mesoveliidae		Ecnomidae		Moths	
Psammaspididae		Plecoptera		Micronectidae		Glossosomatidae		Crambidae [ex-Pyralidae]	
Amphi-Chiltoniidae [Ex. Ceinidae]		Austroperlidae		Naucoridae		Helicophidae			
Amphi-Corophiidae		Eustheniidae		Nepidae		Helicopsychidae		OTHER TAXA	
Amphi-Eusiridae		Gripopterygidae		Notonectidae		Hydrobiosidae			
Iso-Sphaeromatidae		Notonemouridae		Ochteridae		Hydropsychidae			
Iso-Corallanidae		Diptera		Pleidae		Hydroptilidae			
Atyidae		Athericidae		Saldidae		Leptoceridae			
Palaemonidae		Blephaceridae		Veliidae		Odontoceridae			
Parastacidae		Ceratopogonidae		Coleoptera		Philopotamidae			
Parathelphusidae		Corethrellidae		Carabidae		Philorhreithridae			
Trogloplacidae		Chaoboridae		Chrysomelidae		Polycentropodidae			
Mollusca		Chironomidae		Curculionidae		Psychomyiidae			
Ancylidae [part Planorbidae]		s-f Chironominae		Dytiscidae		Tasimiidae			
Bithyniidae		s-f Orthoclaadiinae		Elmidae		Stenopsychidae			

Background information on sampling bores for stygofauna

Version: February 2018

3. Background information on sampling bores for stygofauna

3.1. Purpose and scope

This document provides background information on sampling bores for stygofauna (subterranean aquatic fauna).

3.2. Associated documents

Biological assessment: Sampling bores for stygofauna

3.3. Introduction

Stygofauna, also known as subterranean aquatic fauna, are aquatic fauna that live part or all of their lives in groundwater systems such as aquifers or underground caves. Stygofauna are found in aquifers and caves, inhabiting the water filled pore spaces, voids, cracks and fissures of most rock types including sandstones, laterites, calcretes and basalts, in natural and modified springs and unconsolidated sediments (Humphreys 2006, Humphreys 2008).

The majority of stygofauna are crustaceans. Other stygofauna taxa include mites, worms, snails, insects and fish. These animals live in the dark where primary production is limited, and because of this, some stygofauna rely on organic matter introduced into their environment, predominantly from seepage of water from the surface. Other stygofauna are predators or use bacterial biofilms as a food source (EPA 2012 and references cited within). Some stygofauna species are adapted to living their entire life in total darkness. A group of stygofauna call stygobites live exclusively in groundwater and are characterised by the loss or severe reduction of eyes and pigment (**Figure 44**).



Figure 44: Examples of stygofauna (a) Acarina (mite) (photo credit A. Prior, DNRM) and (b) Syncarida (photo credit A. Steward, DES)

Stygofauna are a key part of Australia's biodiversity and tend to have a high degree of endemism (Humphreys 2006). They are potentially impacted by groundwater withdrawal and changes in groundwater quality (Nevill et al. 2010, Hartland et al. 2011). Stygofauna in Queensland have been described at depths of up to 60m below ground, at electrical conductivities above 50,000 μ S/cm and in both acidic (pH 3.5) and alkaline (pH 10.3) environments (Schultz et al. 2013, Glanville et al. 2016). The Queensland Government has provided a guideline for the environmental assessment process of stygofauna and contains information on the design of stygofauna sampling programs. This can be found at: <https://publications.qld.gov.au/dataset/subterranean-aquatic-fauna>.

3.4. Sampling stygofauna

There are two recommended sampling methods for stygofauna—netting and pumping (see Sampling bores for

stygofauna document). Netting is a passive sampling method that collects animals residing within the bore casing (**Figure 45**). Pumping is an active sampling method that collects groundwater and fauna from within the bore casing and the surrounding aquifer substrate. As the pump actively draws water and fauna into the bore, it effectively samples a larger area outside the bore, but sampling time can be longer than netting.

The choice of groundwater sampling equipment, particularly the pump that is used, is important as to minimise damage to the animals being collected. Although most pumps can be used, impeller driven pumps such as electric submersible pumps are more likely to damage fauna during collection. The pump used should be able to deliver water to the surface at a rate >10L/min from a water table 40m below ground to ensure that animals are drawn in from the surrounding aquifer (EPA 2007).



Figure 45: Example of a net used to sample stygofauna, with collection vial attached to bottom of net

3.5. Stygofauna database

The Queensland Herbarium manages a Queensland Government Subterranean Aquatic Fauna Database that contains a collection of stygofauna locations and species throughout Queensland. Upon completion of projects involving stygofauna sampling, results should be sent to the Queensland Herbarium to ensure the database is kept up to date (Queensland.Herbarium@qld.gov.au). Data provided must be in a suitable format as detailed in the DSITI Guideline for the Environmental Assessment of Subterranean Aquatic Fauna (DSITI 2014).

This database is available upon request to the Queensland Herbarium (Queensland.Herbarium@qld.gov.au), and will be made publicly available in the future.

3.6. References and additional reading

DSITI 2014, Guideline for the Environmental Assessment of Subterranean Aquatic Fauna, Department of Science, Information Technology and Innovation, Queensland Government, viewed 26 July 2016, <https://publications.qld.gov.au/dataset/subterranean-aquatic-fauna>.

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EPA (Environmental Protection Authority) 2012, A review of subterranean fauna assessment in Western Australia, Available from:

<http://edit.epa.wa.gov.au/EPADocLib/Disc%20paper%20OEPA%20subterranean%20fauna%20v2%200%20final%20Mar%202012.pdf>.

EPA (Environmental Protection Authority) 2013, Environmental assessment guideline for Consideration of subterranean fauna in environmental impact assessment in Western Australia, EAG 12, Available from: <http://edit.epa.wa.gov.au/EPADocLib/EAG12%20Subterranean%20fauna.pdf>.

Glanville, K, Schulz, C, Tomlinson, M, Butler, D 2016, 'Biodiversity and biogeography of groundwater invertebrates in Queensland, Australia', *Subterranean Biology*, 17, 55-76.

Hancock, PJ, Boulton, AJ, Humphreys, WF 2005, 'Aquifers and hyporheic zones: Towards an ecological understanding of groundwater. The Future of Hydrogeology', *Hydrogeology Journal* 13, 98-111.

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Schultz, C, Steward, A, Prior, A 2013, 'Stygofauna presence within fresh and highly saline aquifers of the Border Rivers region in southern Queensland'. *Proceedings of the Royal Society of Queensland*, 118, 27-35.

WetlandInfo 2013, Aquifers and caves, Department of Environment and Heritage Protection, Queensland Government, Queensland, viewed 26 July 2016, <http://wetlandinfo.ehp.qld.gov.au/wetlands/ecology/aquatic-ecosystems-natural/aquifers-caves/>.

Sampling bores for stygofauna

Version: February 2018

4. Sampling bores for stygofauna

4.1. Purpose and scope

This document describes the procedure for sampling stygofauna from groundwater monitoring bores using both netting and pumping methods. This method is an adaptation of a sampling protocol developed by ALS Water Resources Group (Hancock & Bennison 2011).

4.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping including taking of field photographs and videos*

Physical and chemical assessment: Guidance on the sampling of groundwaters

Biological assessment: Background information on sampling bores for stygofauna

4.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

4.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals for more information on requirements.

4.5. Skills, training and experience

Skills, training and/or experience required to understand and/or undertake this method include training and experience in groundwater sampling.

4.6. Equipment

See Appendix 1 for example equipment checklist.

4.7. Procedure

4.7.1. Preparation for sampling

- Ensure the correct sampling method (netting and/or pumping) has been chosen to meet the objectives of the study.
- Check that the stygofauna nets are free of holes.
- Ensure that pump for groundwater sampling is in good working condition.
- Download DNRM bore cards for the groundwater monitoring bores you will be sampling. Bore cards can be located using the Google Earth™ and Queensland Globe (<https://data.qld.gov.au/maps-geospatial/qld-globe>). Load the Inland waters globe to find details of groundwater monitoring bores. Click on the bore of interest to download the bore card from Google Earth™.

4.7.2. Sampling

1. Record the bore number, bore diameter, bore depth, height of collar, whether casing extends for the entire length of bore, and whether the bore is screened. This information should be provided on DNRME bore cards. If a bore card is not available, measure as a minimum the depth of the bore and the water depth.
2. Take and record a GPS reading for the bore.
3. Take photos of bore and surroundings as a record of local vegetation and the immediate landscape.
4. Measure and record depth to water table and depth to bottom of the bore using water level meter.
5. If ground water quality samples are to be collected these should be collected after the stygofauna have been sampled (See Groundwaters document).

4.7.2.1. Netting

1. Attach the collection vial to bottom of the net (**Figure 46a**), and the net to a fishing reel.
2. Lower the net to the bottom of the bore (**Figure 46b**) using a fishing reel.
3. Once the net has reached the bottom of the bore, raise the net up and down to dislodge any fauna attached to the bottom of the bore. The net should be drawn up and down a distance of approximately 30cm and a total of four times.
4. Reel the net up in a smooth and steady motion (~10-20cm/sec) to avoid a bow wave and losing any fauna captured.
5. Place a 50µm sieve into the bottom of a plastic bucket. Once the net is clear of the bore, remove the collecting vial and pour the contents into the 50µm mesh sieve in the bucket. Ensure the net does not touch the ground.
6. Hold the net over the sieve and wash using water from a wash bottle (**Figure 46c**).
7. Repeat steps 1 to 5 six times in total.
8. When reeling in the line the final time at a bore, wipe the line with a cloth as it is reeled in to wipe off any fauna that may be stuck to it.
9. Rinse the net, vial and sinkers over the sieve.
10. Tilt the sieve and wash the contents of the sieve into a sample jar (**Figure 46d**). Preserve the sample with 100% ethanol and stain with a small quantity of Rose Bengal stain.

Note: For genetic studies, Rose Bengal should be avoided as it binds to genetic material.

11. Record bore number, water level depth, collection date, sample number and sample type (i.e. net or pump) on a label and add label to jar.
12. Wash the stygofauna net, collection vial and weights in Decon90 solution and then rinse thoroughly in tap water. Nets should be allowed to dry between sites.
13. Ensure bore cover is replaced and locked as found.



Figure 46: Sampling stygofauna using a net and reel

4.7.2.2. Pumping

1. Set up the groundwater sampling equipment as per manufacturer's instructions.
2. Note: Although most pumps can be used, impeller driven pumps such as electric submersible pumps are more likely to damage fauna during collection and this should be avoided if possible.
3. Feed the sampling hose into the bore until it reaches the bottom of the bore casing. Then lift hose so that it sits approximately 2m above the bottom of the bore.
4. Set out three rows of ten x 10L buckets, each row equivalent to a 100L sample (**Figure 47a**). Buckets with spouts are recommended, and it is helpful to have three different coloured buckets—one colour for each row. If the area surrounding the bore is vegetated, clear the vegetation to provide flat surface. A weed trimmer/brush cutter maybe required for thick vegetation.
5. Start the pump and hold the end of the hose close to the bottom of the first bucket ensuring that it is below water level as the water flows in. This will reduce aeration for physico-chemical measurements.

6. Fill buckets sequentially and try to minimise splashing and overflows.
7. Use multi-probe to measure physico-chemical parameters from first bucket and then every 50L i.e. every five buckets.
8. Once the water quality parameters measurements have been recorded, sieve the contents of each bucket through a 50µm mesh sieve (**Figure 47b**). Gently swirl the contents of the bucket so that organic matter and fauna are suspended, then carefully pour through the sieve. Capture the sieved water in another bucket and then transfer back to original bucket. Elutriate and sieve twice more.
9. After sieving the first 10 buckets (i.e. 100L), transfer the sieve contents into a sample jar and preserve with 100% ethanol.
10. The sample jar should be labelled with a permanent marker label and a pencil label on waterproof paper should be placed in jar.
11. Repeat steps 8 and 9 for second and third 100L, so that there are three x 100L samples per bore site.
12. Record bore number, water level depth, collection date, sample number and sample type (i.e. net or pump), and sub-sample details (i.e. sub-sample 0-100L, 100-200L, 200-300L) on a label and add label to jar. Record these details in a notebook or equivalent.
13. Add a small amount of Rose Bengal to each sample jar.

Note: for genetic studies, Rose Bengal should be avoided as it binds to genetic material impacting on genetic analysis.

14. Water quality samples (e.g. Nutrients, Dissolved Organic Carbon, pesticides etc.) should be collected straight from the hose once stygofauna sampling has finished (i.e. after 300L has been collected in sample buckets).
15. Sample bottles must be labelled and the sample name, site, data and time and sampler name must be recorded in a notebook or equivalent.
16. After sampling is completed, remove hose from the bore and pump and empty it of water. Pump a solution of Decon90 through the hose to decontaminate it, then pump thoroughly with tap water.
17. Wash the outside of the hose and wipe dry with a towel to prevent grass and dirt sticking to the hose and contaminating the next bore.
18. Rinse and dry sieve.
19. Ensure bore cover is replaced and locked as found.
20. Chain of Custody (CoC) documentation should be filled out.



(a)

(b)

Figure 47: Sampling stygofauna using a pump and buckets

4.8. References and additional reading

EPA (Environmental Protection Authority) 2007, Guidance for the Assessment of Environmental Factors: Sampling methods and survey considerations for subterranean fauna in Western Australia, Guidance Statement 54a, draft, Available from: http://www.epa.wa.gov.au/EPADocLib/2543_GS54a30708.pdf.

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Hancock, P, and Steward, A 2004, Pioneer Groundwater Biota Pilot Study. Review of Sampling Methods for Groundwater Fauna and Bacteria, Aquatic Ecosystems Technical Report No. 49, Queensland Department of Natural Resources and Mines, Indooroopilly, Brisbane, Queensland.

Appendix 1

Table 1 Equipment checklist

Equipment	✓
Inertial pump	
30m hose	
Stygofauna sampling net (50µm mesh) with fishing rod and reel	
Groundwater level monitoring tape	
Vials and sinkers to attach to net	
50µm mesh sieve	
Squeeze bottle for water and ethanol	
Sample bottles and jars	
Waterproof markers and pencils	
Thirty 10L buckets	
Water quality meters	
5L containers for Decon90	
Notebook or field sheets	
Safety equipment	

Background to fish sampling and index calculation

Version: February 2018

5. Background to fish sampling and index calculation

5.1. Purpose and scope

The purpose of this document is to provide some general information on fish sampling and index calculation.

5.2. Associated documents

- Biological assessment:
- Sampling fish communities using fyke nets
- Sampling fish communities using bait traps
- Sampling fish communities using gill nets
- Sampling fish communities using electrofishing
- Sampling fish communities using seine nets
- Sampling fish communities using cast nets
- Fish holding, identification and measurement of length and weight
- Fish collection and dissection for the purpose of chemical analysis of tissues

5.3. Introduction

Fish communities are useful indicators in assessing aquatic ecosystem health (Kennard et al. 2001) because they are sensitive to changes in water quality and habitat structure which may occur as a consequence of either natural causes or anthropogenic activities (Pidgeon 2004). Sampling of fish communities both spatially and temporally can provide valuable information about any natural and/or human disturbances. Comparisons of fish community structure and abundance data collected across sites (including control and/or reference sites) and over time (if possible) can assist in identifying potential impacts from human activities such as water pollution events, poor land use practices, alteration in stream flow and connectivity, reduced riparian vegetation and sediment aggradation on the stream bed.

Fish communities have the following attributes, making them useful indicators for assessing aquatic ecosystem health (Kennard et al. 2001):

- fish are a taxonomic group commonly found in nearly all aquatic environments
- fish are relatively long-lived and mobile, thereby able to reflect conditions over a broad temporal and spatial scale, providing valuable information about the overall catchment health
- a variety of species make up the fish community over a range of trophic levels
- fish are consumed by humans, and consequently there is often a public interest in fish communities
- fish are useful in detecting potential contaminants through tissue analysis (see Biological assessment—Fish collection and dissections for further information)
- fish are easily collected using a variety of fish sampling methods, and it is possible to sample, identify and release the individuals back into the water unharmed.

There are a variety of methods for sampling fish communities, and designing a fish sampling program will need to take into consideration a number of factors, such as:

- the aim of the sampling
- habitats available for sampling
- amount of time available for sampling
- experience of the sampler/s
- equipment available
- necessary permits and authorities (e.g. General Fisheries Permit, Animal Ethics approval), and associated restrictions.

5.4. Fish sampling methods

Fish sampling methods can be either active or passive, with active gear moved through the water column to capture fish, whereas passive gear is set stationary within the water column to collect fish swimming into it. Each fish sampling technique has advantages and disadvantages, and the use of each will depend upon the environment to be sampled.

Active sampling techniques include electrofishing, seine netting and cast netting. These techniques allow the sampler to actively target communities and provide immediate results. These are ideal for rapid assessments; however, each method has some limitations. For example, electrofishing is less effective in high conductivity waters (e.g. $>1000\mu\text{s}/\text{cm}$), and seine netting is difficult in areas with many snags in the water.

Passive fish sampling techniques include fyke nets, gill nets and bait traps. These methods allow nets/traps to be set in targeted habitats, and are designed to be left in the water and collected after a predetermined amount of time. These methods may be more time-consuming as often they require the sampler to return to a site after a set period of time to collect the net and process the fish catch.

Fish sampling methods vary in efficiency across fish species, and environmental characteristics, e.g. water turbidity, depth, flow. Often, a variety of sampling techniques may be needed to address the aim of sampling.

Detailed information on each fishing method can be found in the documents listed in Section 2 – Associated Documents. Information on the permits and approvals required for conducting a fish survey can be found in the Sampling design and preparation—Permits and approvals document.

5.5. Fish indices

Fish indices have been developed to aid in interpretation of sampling results, particularly when sampling has been conducted in a quantitative manner. There are currently three fish indices prescribed as water quality objectives (WQOs) under the Environmental Protection (Water) Policy 2009 (EPP Water), which falls under the Environmental Protection Act 1994. WQOs are measures established to protect the environmental values of the waterway, and apply to receiving waters. It should be noted that WQOs are not currently available for every catchment in Queensland¹⁴.

The three fish indices currently prescribed as WQOs under the EPP Water are:

- Percentage of Native Species Expected (PONSE)
- Ratio of Observed to Expected species ratio (O/E50) and,
- Percent of Alien Fish individuals.

Each of these indices is described below.

5.5.1. Percentage of Native Species Expected (PONSE)

The Percentage of Native Species Expected (PONSE) indicator is the number of native fish species observed at a site as a percentage of the number of native fish species expected to occur at a physically similar site under minimally-disturbed conditions. Kennard et al. (2001) found that the number of native fish species declines with increasing level of disturbance, and the PONSE indicator can reflect a variety of sources of disturbance at a range of spatial and temporal scales. Only the total number of different native species found at a site compared to the expected number is used to calculate this index; the identities of the native species present are not taken into account.

To calculate the PONSE index, a numeric model is used to determine the expected number of native fish species, usually based on landscape (e.g. site elevation, distance from river mouth, distance from source) and/or habitat variables (e.g. stream width, depth). PONSE is expressed as a percentage, with a score of 100% indicating the number of native fish species observed is the same as expected at a site under minimally disturbed conditions; and a score close to 0% suggesting there is a high level of disturbance at the site. The current WQOs for the PONSE index are 100%.

Background on the development of the original model for PONSE can be found in Kennard et al. (2001, 2006a). Details of data requirements and model predictions may be obtained via e-mail from water.data@qld.gov.au.

5.5.2. Ratio of Observed to Expected native species (O/E50)

The ratio of Observed to Expected native species (O/E50) is the ratio of native fish species observed at a site against the native fish species expected to occur with $\geq 50\%$ probability of occurrence at a physically similar site under minimally-disturbed conditions. The O/E50 ratio varies from the PONSE index in that it takes into account, at least in part, the identity of the native fish species caught and predicted to occur, rather than just the number of different native fish species. This means changes in species composition at a site from a minimally disturbed site

¹⁴ Further information can be found at <http://www.ehp.qld.gov.au/water/policy/>

can be assessed. As with the PONSE index, a numeric model is used to determine the expected native fish species at a site.

O/E50 scores are expressed as a ratio, with a score closer to one suggesting the fish assemblage observed at the site is close to what is expected at the site under minimally-disturbed conditions; and a score closer to zero suggesting the fish assemblage is different to what is expected at a minimally disturbed site, possibly due to anthropogenic disturbance. Kennard et al. (2001) found that low O/E50 scores were strongly associated with poor in-stream habitat, and disturbances due to land use, changes in water chemistry and channel degradation. The current WQOs for the O/E50 index are set at one. Details on the development of the original model used to calculate the O/E50 can be found in Kennard et al. (2001, 2006b), and a description of an improved model has been provided by Rose et al. (2016b). Details of data requirements and model predictions may be obtained via e-mail from water.data@qld.gov.au.

5.5.3. Percentage of alien fish individuals

The percentage of alien fish index is the number of alien¹⁵ fish individuals expressed as a percentage of the total number of individuals caught. Research conducted by Kennard et al. (2001, 2005), and Rose et al. (2016c) indicated that water bodies affected by human activity and modification are more susceptible to invasion by alien fish species, and conversely, often less suitable as habitat for sensitive native fish species. As such, fewer native species and more alien species may be expected (Kennard et al. 2001, Rose et al. 2016c).

The current WQOs for the proportion of alien fish is zero.

5.6. References and additional reading

Kennard, MJ, Harch, BD, Arthington, AH, Mackay, SJ and Pusey, BJ 2001, 'Freshwater fish as indicators of ecosystem health', in MJ Smith and AW Storey (eds), *Design and Implementation of Baseline Monitoring (DIBM3): Developing an Ecosystem Health Monitoring Program for Rivers and Streams in Southeast Queensland*, South-East Queensland Regional Water Quality Management Strategy, unpublished report, Brisbane, pp. 9.1-9.61.

Kennard, MJ 2005, A quantitative basis for the use of fish as indicators of river health in eastern Australia, Unpublished PhD thesis, Faculty of Environmental Sciences, Griffith University, Brisbane, Australia.

Kennard, MJ, Arthington, AH, Pusey, BJ and Harch, BD 2005, 'Are alien fish a reliable indicator of river health?' *Freshwater Biology* 50 (1), 174-193.

Kennard, MJ, Harch, BD, Pusey, BJ and Arthington, AH 2006a, 'Accurately defining the reference condition for summary biotic metrics: a comparison of four approaches', *Hydrobiologia* 572 (1), 151-170.

Kennard, MJ, Pusey, BJ, Arthington, AH, Harch, BD and Mackay, SJ 2006b, 'Development and application of a predictive model of freshwater fish assemblage composition to evaluate river health in eastern Australia', *Hydrobiologia* 572 (1), 33-57.

Pidgeon, B 2004, A review of options for monitoring freshwater fish biodiversity in the Darwin Harbour catchment, Report prepared for Water Monitoring Branch, Natural Resource Management Division, Department of Infrastructure, Planning and Environment, Department for the Environment and Heritage Supervising Scientist.

Rose, PM, Kennard, MJ, Sheldon, F, Moffatt, DB, Butler, GL 2016a, 'A data-driven method for selecting candidate reference sites for stream bioassessment programs using generalised dissimilarity models', *Marine and Freshwater Research*, 67, 440-454.

Rose, PM, Kennard, MJ, Moffatt, DB, Sheldon, F, Butler, GL 2016b, 'Testing three species distribution modelling strategies to define fish assemblage reference conditions for stream bioassessment and related applications', *PLoS ONE*, 11 (1).

Rose, PM, Kennard, MJ, Moffatt, DB, Butler, GL, Sheldon, F 2016c, 'Incorporating species losses and gains into a fish-based index for stream bioassessment increases the detection of anthropogenic disturbances', *Ecological Indicators*, 69, 677-685.

¹⁵ Alien species are fish species originating from outside of Australia. Species translocated within Australia are not classified as alien fish.

Sampling fish communities using electrofishers

Version: February 2018

6. Fish holding, identification and measurement of length and weight

6.1. Purpose and scope

This document describes how to hold, identify and measure the length and weight of fish after they have been collected from nets/traps.

6.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

6.3. Health and safety

Before following the methods contained in this document, a detailed risk management (identification, assessment, control and review of hazards and risks) must be undertaken. All work carried out must comply with Queensland Work Health and Safety legislative obligations.

6.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See Permits and approvals document for more information on requirements.

6.5. Skills, training and experience

Skills, training and or experience required to understand and/or undertake this method include:

- ability to identify fish to species level
- prior experience in the measurement of fish.

6.6. Equipment

See Appendix 1 for example equipment checklist.

6.7. Procedure

6.7.1. Temporary holding of fish

1. Place captured fish into a holding container (e.g. Nally® bin). This container is to be filled with water sourced from the site. If the fish catch is large, change water often during processing.
2. Attach a suitable aerator to the container if required, placing the air stone into the water to ensure water is adequately oxygenated. More than one aerator may be required for large volumes of fish and/or in warmer temperatures. Wherever possible, keep the holding container in the shade and/or partially resting within the waterway to take advantage of the thermal mass of the larger water body.
3. Undertake all processing immediately in order to minimise stress to fish.

Note:

- Utilise coarse and/or fine sieves as required to rinse debris from small fish when processing fish from a holding container containing large amounts of detritus.
- Ensure captured fish are monitored continuously for signs of stress and oxygen depletion (e.g. loss of equilibrium or air gulping) and rectify by partial water changes or release of the fish.

6.7.2. Identifying and measuring fish

1. Wet hands prior to touching fish to avoid damaging sensitive mucous layers. Handling should be minimised to avoid unnecessary stress and/or mortality.
2. Begin processing by removing larger individual fish first. These are easier to retrieve and removal will reduce stress on smaller fish in the holding container.
3. Remove the fish from the holding container and identify. Record the species in a notebook or equivalent. Supporting resources such as keys, books, photographs may be used. If unable to identify a species, preservation of specimens may be required after the individual has been measured (see Section 6.7.4).
4. Place the fish on the measuring board, with the snout positioned at 0cm. Measure either standard length, total length or fork length (as described below) in millimetres. Record the length in a notebook or equivalent. **Note:** The most important factor in measurement is consistency; i.e. if standard length was used at the site in the past to measure a golden perch, then it must be consistently used as the length measure for that species. Being consistent ensures that measurements are comparable over time.
 - **Standard length** is the length from snout to the tail-end of the vertebral column (the hypural crease) (**Figure 48**). The hypural crease can be located by holding the fish against a flat surface and gently bending the tail upwards and forward such that a crease (the hypural crease) becomes visible. Standard length provides the most reliable length measurement because tail fins (caudal fins) are often damaged, making it difficult to determine total or fork length.
 - **Total length** is the measurement taken from the tip of the snout to the end of the caudal fin (**Figure 48**).
 - **Fork length** is the measurement taken from the tip of the snout to the centre of a concave tail (**Figure 49**).
5. Weigh the individual fish following the manufacturer's instructions for the scales in use.
6. Place the scales in a stable, sheltered location. If required place the fish into a tray, ensuring the scales read zero prior to the fish being placed on the scales.
7. Remove as much excess water from individual fish as practicable.
8. Place fish on the scale and measure in grams. Record the fish weight in a notebook or equivalent.
9. Return fish to the water as quickly as possible once measurements and recordings have been made.

Note:

- When processing fish, one person should be dedicated to identifying and measuring fish and another person should be dedicated to taking accurate notes on appropriate data sheets and assisting where required.
- Where catch volumes are very high, it may be necessary to subsample from the total catch in order to avoid long processing times and reduce fish stress and/or mortality. The number of individuals sub-sampled will depend on the aims of the particular study. For example, Kennard et al. (2011) suggest a sub-sample of 30 individuals from each species.

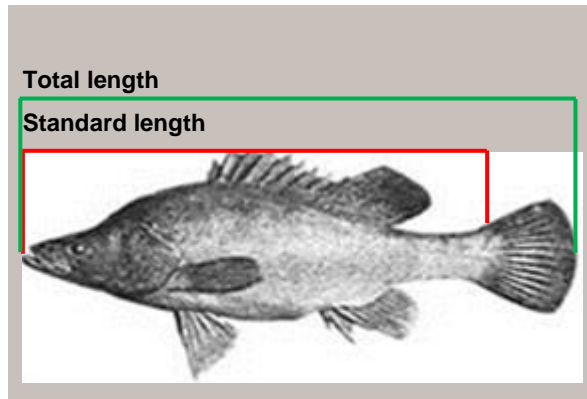


Figure 48: Golden perch (*Macquaria ambigua*): standard length is measured from snout to the tail-end of the vertebral column. Total length is measured from tip of snout to tip of convex tail.



Figure 49: Hardyhead (*Atherinosoma microstoma*): fork length is measured from tip of snout to centre of concave tail.

6.7.3. Releasing fish

1. Release all native fish back into the water they came from. If fish cannot be identified, representative specimens may be preserved for later identification (as outlined below).
2. Euthanize noxious fish as required by the General Fisheries Permit and Animal Ethics approval and dispose of appropriately. Consult the Queensland fisheries website for the latest information on policies and legislation regarding the release of noxious fish.

6.7.4. Preserving fish

1. If fish are to be taken from site (for identification or further analysis), euthanize fish in accordance with your General Fisheries Permit and Animal Ethics Approval.
2. Store the fish in an appropriate container (if preserving in liquid) or double-lined zip-locked bags (if freezing). Label these with details including site, fishing method, identification and preservation type. Examples of preservation include 70% ethanol or 10% formalin for identification and freezing for contaminant analysis. Record any fish collected in a notebook or equivalent.

6.8. References and additional reading

Allen, GR, Midgley, SH and Allen, M 2002, Field guide to the freshwater fishes of Australia, Western Australia Museum, Perth, 394 pp.

Jearld, Jr A 1983, Active Fish Capture Methods, in LA Nielsen and DL Johnson (eds), Fisheries Techniques, American Fisheries Society, Southern Printing Company Inc. Blacksburg, Virginia.

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Pusey, B, Kennard, M and Arthington, A 2004, Freshwater fishes of north-eastern Australia, CSIRO Publishing, Collingwood, Victoria.

Sampling fish communities using electrofishers

Version: February 2018

7. Sampling fish communities using electrofishers

7.1. Purpose and scope

This document provides the procedure that can be used across the different forms of electrofishing (e.g. backpack, shore-based and boat-mounted units). It broadly describes checking equipment, preparation for sampling and the basic steps of electrofishing. Due to the variation in electrofishing equipment and purposes, it is important to refer to the manufacturer's manual, Australian Code of Electrofishing Practice and relevant background information for specific details on the use of electrofishing equipment.

7.2. Associated documents

Sampling *design and preparation*:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological Assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

7.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Australian Code of Electrofishing Practice (NSW Fisheries 1997) (hereafter the Code), and Queensland Work Health and Safety legislative obligations.

7.4. Permits and approvals

Any staff member involved in electrofishing must have the approval of their manager, who should ensure the electrofishing operations are compliant with relevant legislation.

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

If a boat is to be used for research and/or commercial purposes, consideration must be given as to whether a Certificate of Survey or Marine Safety (Scientific research and educational activities) Exemption is required.

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See *Permits and approvals* document for more information on requirements.

7.5. Skills, training and experience

All staff involved in electrofishing must have (as a minimum standard) skills to electrofish safely.

Electrofishing team leaders must meet the standard of a Senior Operator with respect to the type of electrofishing equipment to be used, as defined in the Code. This includes the requirement of 50 hours of 'supervised electrofishing' to qualify as a Senior Operator. 'Supervised electrofishing' is based on active fishing time, i.e. excluding preparation and travel time. Note that the qualification time must be fully documented.

No person is to operate electrofishing equipment unless they meet the standard for a Senior Operator, or, for training purposes, work under the direct close supervision of a Senior Operator. For boat-based electrofishing, there must be one Senior Operator on the boat. Any other team members on board can be in training.

It is highly recommended that Senior Operators undertake theoretical training with an authoritative external organisation such as the US Fish and Wildlife Service (<http://nctc.fws.gov/courses/csp/csp2c01/resources/>) and/or

Smith-Root Inc. as part of their initial training. It is also recommended that Senior Operators take time to periodically refresh their theoretical knowledge and/or practical skills, especially if they have not electrofished for a period of six months or more.

Staff members assisting the team leader need to complete:

- a detailed briefing on dry land with the electrofishing team leader, who must demonstrate operation of equipment and provide safety information
- a “wet induction” with the electrofishing team leader that involves active fishing solely for demonstration (as opposed to data collection) purposes and highlights safety requirements.

All staff involved in electrofishing must hold a current Senior First-aid Certificate or equivalent, including CPR, and have written approval from a medical doctor that states that they are fit for electrofishing. This may include an ECG report.

If a boat is to be used, the skipper must be sufficiently experienced with the size of the boat being used and the prevailing river or lake conditions. It is recommended that the skipper holds a current Queensland Recreational Shipmaster’s Licence.

At least one staff member must have the ability to identify local fish to species level.

7.6. Equipment

Equipment will depend upon the type of electrofishing to be undertaken. See Appendix 1 and 2 for an example equipment checklist.

7.7. Procedure

7.7.1. Determine fishing protocol

There are many fishing protocols that could be used, and these depend upon the aim of the sampling, site characteristics and electrofishing equipment used. Some considerations are outlined below when determining your electrofishing protocol.

7.7.1.1. Aim of sampling

Understanding the purpose of the sampling will assist in determining the fishing protocol and the methods to be undertaken during electrofishing sampling.

If the aim is to conduct a fish community composition survey, then fishing effort is typically timed (e.g. five minute shot time), with the number of shots depending on the size of the site. Electrofishing may be conducted across alternate banks and mid-channel to cover all habitat types, or may be divided into habitats, transects or sample reaches. For example, sampling may be conducted with runs both parallel and perpendicular to the bank to capture fish from various habitats (e.g. in the edge and mid-water). Other examples may be to sample for a fixed time per shot in homogenous habitats at each site, to assist in standardisation, or to attain full coverage of a certain section of stream, denoted by length of stream section or area fished.

If the aim is to target a particular fish species (e.g. golden perch), sampling methods will be designed to target specific habitats for the particular fish species (e.g. snags in deep pools). More sampling effort will be placed on habitats known to be suitable for the particular species.

Recording measures of fishing effort (e.g. ‘power-on’ time, fishing time, size of fished area) is important for all electrofishing sampling protocols, as it allows some standardisation across sites and over time to provide accurate comparisons in community composition (e.g. catch per unit effort), or calculation of effort when targeting species.

7.7.1.2. Site characteristics

Knowing the site/s to be sampled will assist in determining your sampling protocol. The size of the water body, depth to be sampled, habitat types, flow of the water and approximate water conductivity will help in determining the equipment to be used, and the sampling methodology.

7.7.1.3. Equipment type

There are many different electrofishing units (e.g. backpack, boat-based and shore-based), each having different uses. Equipment is determined by characteristics such as depth/flow of the water and water electrical conductivity. General details for each are below. A combination of types of electrofishing equipment can be used.

Backpack electrofishing is generally used for smaller streams and lakes, typically of a depth less than the operator

crotch height. Strong flows should be avoided using this technique. Backpack electrofishing can only be performed in water of low to moderate salinity (e.g. 10–1500 μ S/cm depending on unit specifications), and therefore, its use is limited in tidal and some lowland waters.

Boat-based electrofishing is used for large rivers and lakes where it is not possible to use backpack electrofishing, or where unsafe to wade (e.g. due to potential presence of crocodiles or strong flows). Boat-based electrofishing can be undertaken in low to high water salinity (e.g. 10–25,000 μ S/cm depending on unit specifications).

Shore-based electrofishing is used in wadeable waters, but has the advantage over backpack electrofishing in that it can be undertaken in waters of moderate to high salinity. However, because they have a higher power output, the safety risk is also greater than backpack-mounted equipment.

7.7.2. Complete pre-departure check of equipment maintenance and testing

The following steps should be undertaken prior to departing to the field.

7.7.2.1. Check maintenance and testing logbook

Ensure the equipment is ready for use and is not requiring any maintenance prior to use. Refer to specific manufacturer's instruction manual for further information on maintenance, and the Australian Code of Electrofishing Practice (NSW Fisheries 1997).

7.7.2.2. Conduct a bench-test of the equipment

The Senior Operator must check the electrofishing equipment (including pole, anode and cathode) to ensure it is in working order prior to field mobilisation. The Senior Operator must also check that the batteries are fully sealed and working, and ensure they are fully charged before undertaking the survey.

7.7.3. Preparation for electrofishing

7.7.3.1. Undertake staff briefing prior to entering the water

This should consist of an overview of electrofishing equipment, including safety features and personal protective equipment, and the safety protocols around electrofishing, such as not touching the water, and clear communication between the operator and the assistant/s. Use the following as a guide:

- Assess the sampling site to be fished before entering the water with the equipment. It may be helpful to mark out the fishing area (e.g. upstream and downstream extent, target habitats) prior to fishing. If backpack electrofishing is to be undertaken, ensure water depth and flow is suitable across the fishing area (i.e. water level not above operator crotch depth, and flow safe to wade in). Similarly, ensure suitable depth is present if using boat-based electrofishing.
- Determine water conditions, such as electrical conductivity, which will affect the nature and range of the electrical field within the water. Knowing the electrical conductivity of the water will assist the Senior Operator in setting the electrofishing output.
- Set electrofisher output. This will be dependent on equipment to be used, and water conditions. The aim of setting the electrofisher output is to use the minimum power required for successful electrofishing. If the power is too low, electrofishing will be unsuccessful as it will not stun fish. If the power is too high, there is potential to harm or kill fish. Because of this, electrofishing settings and/or fishing protocol should be set to minimise harm to the fish while still allowing effective fishing. The Senior Operator must adjust the amount of power by setting the electrofisher output through the equipment controls. If the site and water conditions are well known by the Senior Operator, it may be satisfactory to set the equipment to the output settings known to be ideal. Generally, electrofishing equipment controls include:
 - voltage (V) – this will vary according to the electrical conductivity of the water, with lower conductivity requiring higher voltage (e.g. in headwaters), and higher conductivity requiring lower voltage (e.g. in lowland rivers)
 - frequency (Hz) – the number of electrical pulses (cycles) per second. In general, the larger the fish, the lower the frequency required to stun the fish
 - duty cycle (measured in %, is the ratio of on-to-off time) or pulse width (ms).
- Check safety prior to entering the water:
 - All electrofishers must have an audible alarm when in use.
 - For backpacks, recommended safety features include: a quick-release harness, a magnetic safety switch on the anode pole, a tilt switch and fully sealed dry cell batteries. Additional details can be found in the Australian Code of Electrofishing Practice and/or in the user manual for the specific equipment.

- Boat mounted electrofishers must have a deadman switch requiring two or more operators to engage for power to be applied to water. If one is disengaged, the system will shut off power.
- Electrofishing has specific animal ethics requirements, which will be outlined in the Animal Ethics Approval. The following care should be taken when electrofishing:
 - Check there are no other users in the vicinity, including recreational users and animals (e.g. cattle using the water for drinking/crossing, dogs etc.).
 - Be mindful of potential for larger aquatic vertebrates (platypus, turtles, birds) in the sampling area. Use the lowest power required to successfully catch fish to minimise potential stress and injury.
 - If targeting smaller species, higher power or frequency is often required. However, be cautious as larger fish are more vulnerable to electrofishing than smaller fish. Consider other fishing methods if required (e.g. fyke nets).
 - Avoid the necessity of re-shocking fish by netting immediately and either placing in a bucket, or releasing downstream when recovered. Do not allow fish to be continuously shocked, i.e. inside dip net.

7.7.4. Electrofishing

The electrofishing sampling to be undertaken should be outlined in the sampling plan and will depend on equipment, purpose and specific site characteristics. General points to note for undertaking electrofishing are outlined below.

Note:

- Once fishing begins, the output settings may need to be adjusted by observing fish behaviour and recovery times to ensure the most effective settings are used.
- Nets can be set at the upper and lower ends of a stream section to prevent movement of fish out of the area.
- When using backpack or shore-based electrofishing, one person operates the electrofishing equipment, and the other person either collects specimens using a dip-net and places catch into a bucket if required, or assists operator by recording the species as they are captured. During boat-based electrofishing, the driver maintains their foot on the pedal unless unsafe, and the netter applies power to the water and nets the fish.



Figure 50: Backpack electrofishing being undertaken, with assistant carrying bucket and ready to take notes on catch

The following outlines general steps for electrofishing:

- 1 Carefully enter the water at the downstream extent of the sampling area (via boat or wading). Minimise disturbance to limit scaring fish and to keep turbidity to a minimum. Any other staff/observers must keep a safe distance from the water. For backpack electrofishing, the assistant should stand slightly downstream of the operator, keeping out of the way but ready to capture fish, and taking care not to stand on the cathode behind the operator. Slowly approach the desired habitat for sampling. Take care not to disrupt bed sediment while moving through the water.
- 2 Communicate with sampling team that fishing is about to commence. Assistants should be ready to either

- capture fish or record detail provided by the operator.
- 3 Begin electrofishing, and continue as you move through the habitat being sampled (in accordance with sampling protocol). If using a boat, the boat should approach habitats slowly, and electrofishing can continue while the boat is stationary or manoeuvring within the habitat and while the boat exits the habitat.
 - 4 Immobilised fish should immediately be netted either by the electrofishing operator or the assistant so that the fish are isolated from the electric field.
 - 5 Once captured, fish can either be identified and released downstream upon recovery if there is no chance of recapturing them, or placed in a holding vessel with fresh stream water for catch processing. Allow fish to recover before releasing.
 - 6 Record measures of fishing effort (e.g. 'power-on' time, fishing time, size of fished area).
 - 7 Ensure fishing effort is adequate for the purposes of sampling. For example, effort may be adequate if no new species has been recorded during the last 300 seconds of 'power-on' time.

7.7.5. Catch processing

Specific procedures used to process fish will depend upon the project objectives, and should be completed as quickly as possible to minimise stress. It may be required/possible to identify and release fish (downstream) immediately after capture, and in other instances it may be necessary to collect fish in a bucket during each 'shot', then process these at the end of the shot. Depending upon the purposes of the study, voucher photos, tissue samples/specimens and/or measurements of catch may be required prior to release.

See *Fish holding, identification and measurement of length and weight* document for further information.

7.7.6. Cleaning and maintenance of equipment

All gear must be thoroughly cleaned and dried following use to reduce the risk of spread of biological material between waterways.

Each electrofishing unit must have a detailed instruction manual and gear logbook, in which all maintenance of equipment (among other things) is recorded.

Regular electrical safety checks must be performed by a qualified/experienced service technician on all electrofishing units. Refer to instruction manual for additional details.

Note: no substantial repairs or modifications should be made to any electrofishing equipment unless conducted by a suitably accredited electrical/electronics specialist.

All equipment should be inspected by the Senior Operator for any equipment faults, loose wires or connections etc. following each use.

7.8. References and additional reading

Burkhardt, RW & Gutreuter, S 1995, Improving Electrofishing Catch Consistency by Standardizing Power, North American Journal of Fisheries Management 15, 375-381.

Marshall, C, Negus, P, Winning, M & Johnson, D 2004, Fitzroy Basin Resource Operation Plan Ecological Monitoring and Assessment Program Pilot Study – Part 2 Research Project Indicator Methods, Department of Natural Resources, Brisbane.

National Health and Medical Research Council 2013, Australian code for the care and use of animals for scientific purposes, 8th edition, National Health and Medical Research Council, Canberra.

New South Wales Fisheries 1997, Australian code of electrofishing practice, NSW Fisheries Management Publication No 1.

Pusey, B, Kennard, M & Arthington, A 2004, Freshwater Fishes of North-Eastern Australia, Commonwealth Scientific and Industrial Research Organisation (CSIRO) Publishing, Collingwood, Victoria.

US Fish and Wildlife Service 2000, Principles and Techniques of Electrofishing, US Fish and Wildlife Service, Shepherdstown.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Backpack and shore-based electrofishing: <ul style="list-style-type: none"> • 1 x backpack electrofisher or shore-based electrofisher, including anode, anode pole, cathode • 1 x Pair of long 1000V linesman gloves for each sampling team member • Waders for each sampling team member 	
Boat electrofishing: <ul style="list-style-type: none"> • Boat with outboard motor, stable deck for netter at front of boat and three safety gates around the deck • Electrofishing equipment including generator (2.5V or 5.0V) and control box, anode, anode pole, foot switches. • Rubber boots for each sampling team member 	
Life jackets (if water depth great than 50cm or in specific safety plan)	
Shallow and deep long-handled, non-conductive fish net	
Electrical conductivity meter	
Large holding containers (e.g. Nally® bins)	
Gloves for handling fish (optional)	
Polarized sunglasses	
Fish identification field guide	
Measuring board and scales (if required)	
Electrofisher logbook	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Appendix 2

Table 2 Equipment checklist

Equipment	✓
Waterproof notebook or data sheets for fish sampling and processing	
Clipboard	
Labels	
Permanent marker	
Pencils	
Measuring board, ruler, callipers and/or other measuring devices	
Scales	
Coarse (1cm) and fine (250µm) sieves	
Holding tank (e.g. buckets, Nally® bin or other suitable container)	
Portable aerator (with spare batteries) with air hose and air stone	
Vials/containers for storing fish specimens	
Preservative (e.g. 70% ethanol)	
Fridge/freezer (if required for preserving fish specimens)	
White sorting trays	
AQUI-S®, ice or other euthanasia equipment	

Sampling fish communities using seine nets

Version: February 2018

8. Sampling fish communities using seine nets

8.1. Purpose and scope

This document describes the basic use of a seine net in shallow (wadeable) water. As use of a seine net alone may not provide a representative sample of the local fish community, users should consider using a seine net as one component of a suite of different fishing gear types.

8.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological Assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

8.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

8.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See Permits and approvals document for more information on requirements.

8.5. Skills, training and experience

Skills, training and/or experience required to understand and/or undertake this method include:

- ability to identify fish to species level
- prior experience in the use of seine nets.

8.6. Equipment

See Appendix 1 for example equipment checklist.

8.7. Procedure

8.7.1. Preparation for sampling

- Check the specifications of the seine net (e.g. mesh size, length, depth) to be used are suitable for the fishing conditions, target species and aims of fishing. The use of a seine net is likely to be biased towards catching small fish in shallow waters and the mesh size of the seine net used may strongly influence the size of fish caught. The use of a seine net with a large (>6mm) mesh size may preclude the capture of small species (e.g. *Hypseleotris* spp.) and the use of a small mesh size (<6mm) may slow hauling of a seine net such that fast moving fish (e.g. mullets, grunters) can avoid or escape the net.
- Check the seine net for any tangles or debris caught in the net.

8.7.2. Sampling

- 1 Select a suitable section of stream for seining that is relatively free of woody debris, rocks, algae or plants that may snag the seine net. Ensure there is a flat area to process catch within the section of stream being fished. Note that it may be impractical to seine in many streams due to the presence of snags or fast-flowing water.
- 2 Deploy the seine net perpendicular to the stream bank between two people with each person holding the foot rope against the substrate (usually with their foot) as firmly as practical and holding the head rope (usually in the hand) so that it lies along the water surface. In flowing waters, seining should be undertaken against the direction of water flow (**Figure 51**).
- 3 Haul the extended seine along the desired section of stream as briskly as possible to reduce the likelihood of fish avoiding or escaping the net. The escape of fish can often be reduced by ensuring the seine net maintains a “U” shape and/or the use of a seine net with an integrated pocket. It may be useful to have a person walking behind the net to quickly clear the net from any obstructions during seining, otherwise it may be required to stop seining and start again if the net gets caught.
- 4 Complete seining by having the person holding the shoreward end of the seine stop so that the person holding the other end can swing towards the shore and towards their colleague—the seine net will thus form a loop with both ends at the water edge.
- 5 Retrieve the seine net and catch fish by simultaneously hauling both the foot- and head-rope of both ends of the looped net shoreward. Ensure the foot-rope maintains contact with the substrate to reduce the escape of fish. It may also be useful to ensure the head-rope lies forward of the foot-rope to reduce the likelihood of fish jumping over the net.



Figure 51: A seine net being used to fish a wadeable pool

8.7.3. Catch processing

Unless the fish can be quickly processed directly from the wet net to avoid double-handling, the fish catch should be transferred to a holding container (e.g. Nally® bin) with aerated stream water. See Fish holding, identification and measurement of length and weight document for further information.

8.7.4. Cleaning and maintenance of nets

- Rinse, clean and dry seine net as practical before leaving an assessment site to prevent transfer of pathogens and pest plant and animal species to other locations.

- Ensure net is maintained free of debris and holes.
- Clean/repair as necessary before storage or redeployment.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Seine nets (available in various sizes)	
Buckets	
Large holding containers (e.g. Nally® bins)	
Portable aerator (with spare batteries) with air hose and stone	
Gloves for handling fish (optional)	
Fisheries permit signs	
Fish measuring and sample processing equipment	
Fish identification field guide	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Sampling fish communities using fyke nets

Version: February 2018

9. Sampling fish communities using bait traps

9.1. Purpose and scope

This document describes the basic use of bait traps as part of sampling designed to either provide a representative sample of the local fish community or to target individual species for specific purposes. As use of a bait trap alone may not fulfil these objectives, users should consider using a bait trap as one component of a suite of different fishing gear types.

9.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

9.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

9.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See Permits and approvals document for more information on requirements.

9.5. Skills, training and experience

Skills, training and/or experience required to understand and/or undertake this method include:

- ability to identify fish to species level
- prior experience in the use of bait traps.

9.6. Equipment

See Appendix 1 for example equipment checklist.

9.7. Procedure

9.7.1. Preparation for sampling

- Establish target species and aims of sampling. Bait traps primarily sample smaller fish, such as *Hypseleotris* spp. and setting traps overnight might favour the collection of nocturnal and crepuscular (active at dawn and dusk) species.
- Select appropriate trap type and number. Note that different trap types are available, including opera house and box traps (commonly known as 'funnel' traps) as well as round traps and collapsible traps. The number of traps will be dependent on project aims.

- Select bait/attractant. Commonly used attractants are dried cat food and chemical light attractants (e.g. glow sticks). The chosen attractant should be kept consistent throughout the study. Note that some brands of dried cat food may work better than others and fish species may have individual preferences. Fisheries legislation prohibits the use of certain baits taken from a marine environment unless the bait has been frozen, cooked or preserved. Use of live baits is also restricted in some circumstances.

9.7.2. Deployment of traps

1. Select suitable location/s within the stream for deployment. Traps may be deployed from the bank or by boat or wading.
2. Target in-stream habitat such as snags and aquatic vegetation. Depending upon the aims of sampling it may be necessary to ensure representation of all habitat types and various depths, e.g. bare stream banks and minimum to maximum depth available.
3. Plan to distribute traps so they will be independent of each other.
4. Place a small amount of bait in zipped compartment (**Figure 52**). If chemical light sticks are used as an attractant, crack a chemical light stick and place inside the bait trap. One light stick per trap is sufficient.
5. Ensure rope is attached to the trap and tie the other end to vegetation, a float or a stake (**Figure 53**). This will assist in locating the bait trap, and will also prevent the bait trap from floating away.
6. Ensure all entries into the bait trap are submerged (**Figure 54**). The depth at which they are deployed may depend on available habitat and target species.
7. Record deployment time.



Figure 52: Bait trap showing zip pocket for bait



Figure 53: Securing bait trap to a snag



Figure 54: Setting the bait trap

9.7.3. Retrieval of traps

1. Record retrieval time to assist in calculation of effort measures. If multiple traps are used, retrieve in the order they are deployed.
2. Deposit fish catch into a holding container (e.g. Nally® bin) with aerated stream water. If catch volume is small it may be possible to process directly from the trap.
3. Ensure details about the site are recorded such as the number of traps used and where they were situated, habitat type and depth.

9.7.4. Catch processing

Complete processing as quickly as possible to minimise stress. See Fish holding, identification and measurement of length and weight document for further information.

9.7.5. Cleaning and maintenance of nets

- Rinse clean and dry the bait traps after all of the fish have been released.
- Clean/repair as necessary before storage or redeployment.

9.8. References and additional reading

Gehrke, PC, Brown, P, Schiller, CB, Moffatt, DB and Bruce, A 1995, 'River regulation and fish communities in the Murray-Darling river system, Australia', *Regulated Rivers: Research and Management* 11, 363-375.

Marshall, J, Steward, A, McGregor, G, Marshall, C, Negus, P, Johnson, D, Lobegeiger, J, Davies, P, Harch, B, Winning, M, Choy, S and Cockayne, B 2003, 'Condamine-Balonne Integrated Monitoring Pilot Project: Methods', *Aquatic Ecosystems Technical Report No. 41*, Department of Natural Resources and Mines, Brisbane.

Appendix 1

Table 1 Equipment checklist

Equipment	✓
Collapsible bait traps with rope attached to each	
Bait/attractant	
Holding container (e.g. bucket)	
Portable aerator (with spare batteries) with air hose and stone	
Fisheries permit signs	
Fish measuring and sample processing equipment	
Fish identification field guide	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Sampling fish communities using fyke nets

Version: February 2018

10. Sampling fish communities using fyke nets

10.1. Purpose and scope

This document describes the basic use of a fyke net. As use of a fyke net alone may not provide a representative sample of a fish community, users should consider using a fyke net as one component of a suite of different fishing gear types.

10.2. Associated documents

Sampling *design and preparation*:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological Assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

10.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

10.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See Permits and approvals document for more information on requirements.

10.5. Skills, training and experience

Skills, training and/or experience required to understand and or undertake this method include:

- ability to identify fish to species level
- prior experience in the use of fyke nets.

10.6. Equipment

See Appendix 1 for example equipment checklist.

10.7. Procedure

10.7.1. Preparation for sampling

- Ensure mesh size, hoop size, length, number of wings etc. to be used are suitable for the fishing conditions, target species and objectives of the fishing.
- Select and combine different fyke net types and/or replicates as required to target a range of different fish, e.g. single-winged fyke nets for smaller bodied fish along shallow waterhole margins and large double-winged fyke nets for larger pelagic species in deeper water.

Note: Fyke netting is a passive technique (i.e. fish swim into the net and are not actively targeted). Therefore, some species may not be represented in the sample. For example, some species which are common at the site may be poorly sampled and rare species that move in schools may appear more abundant than they actually are.

10.7.2. Deployment of nets

1. Select a suitable location in the stream for deployment. Avoid locations with high flow, poor accessibility, and the presence of significant amounts of floating and submerged debris. Depth should be considered in terms of health and safety, the potential stranding of trapped fish (in tidal areas) and target species.
2. Deploy net facing either downstream or upstream depending on sampling objectives. Fyke nets can be deployed by boat or by wading.
3. Fix tail-end (cod-end) to a stake or structure such as a tree or rock on the bank or within the water (**Figure 55**). Care should be taken to always maintain an air space in the cod-end to allow captured mammals and reptiles to breathe, either by tethering the net above the water surface or placing a float inside the net.
4. Extend fyke wings out one at a time, spreading out hoops and pulling wings taught, and secure to a stake or other available structure such as a tree (**Figure 55**). Securing one of the wings to the bank will maximise the possibility of catching fish species migrating along waterway margins. The degree of 'spread' of the wings will depend on the characteristics of the location and the aims of sampling. The distance between the end of the wings (distal ends) should be recorded to assist in providing a quantitative measure of sampling effort.
5. Ensure funnels within the hoops are at least partially submerged to allow entry of fish.
6. Record deployment time.

Note: Fyke nets are typically deployed overnight, but if there is a risk of by-catch or target animals being hurt, a shorter time must be considered. If deployed overnight, cod ends must be out of the water.

10.7.3. Retrieval of nets

1. Record retrieval time to assist in calculation of effort measures.
2. Bring wing/s in to first hoop then elevate first hoop clear of the water and shake, ensuring that any captured animals fall down into the next hoop.
3. Continue to shake each further hoop in turn until all animals have been collected into the cod-end of the net.
4. Untie the cod-end and deposit fish catch into a large container with aerated stream water.
5. Complete a comprehensive check of wings and hoops to ensure no animals lodged within any part of the net are missed.

10.7.4. Catch processing

Complete processing as quickly as possible to minimise stress. In situations where catch volumes are very high it may be necessary to subsample from the total catch in order to avoid long processing times and possible fish mortality. See Fish holding, identification and measurement of length and weight document for further information.

10.7.5. Cleaning and maintenance of nets

Rinse, clean and dry fyke net as soon as practical before leaving an assessment site to prevent transfer of pathogens and pest plant and animal species to other locations.

Ensure net is maintained free of debris and holes.

Clean/repair as necessary before storage or redeployment.



Figure 55: Fyke net set (photo credit: Hydrobiology Pty Ltd)

10.8. References and additional reading

Gehrke, PC, Brown, P, Schiller, CB, Moffatt, DB, Bruce, A 1995, River regulation and fish communities in the Murray-Darling river system, Australia, *Regulated Rivers: Research and Management*, 11, 363-375.

Gilligan, D, Schiller, C 2003, Downstream transport of larval and juvenile fish in the Murray River, NSW Fisheries Final Report Series No. 50, NSW Fisheries, Narrandera.

Marshall, J, Steward, A, McGregor, G, Marshall, C, Negus, P, Johnson, D, Lobegeiger, J, Davies, P, Harch, B, Winning, M, Choy, S, Cockayne, B 2003, Condamine-Balonne Integrated Monitoring Pilot Project: Methods, Aquatic Ecosystems Technical Report No. 41, Department of Natural Resources and Mines, Brisbane.

Sternberg, D, Cockayne, B, Schmarr, D, Duguid, A, Mathwin, R, McNeil, D 2014, Lake Eyre Basin Rivers Assessment (LEBRA) 2012/13 Monitoring Report, A report to the Department of the Environment, Canberra, ACT.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Fyke net/s (number and size specifications depend upon sampling design)	
Net mending kit (including extra net mesh)	
Boat & outboard (for setting nets in deeper water)	
Coarse & fine strainers	
Large holding containers (e.g. Nally® bins)	
Portable aerator (with spare batteries) with air hose and stone	
Gloves for handling fish (optional)	
Fisheries permit signs	
Fish measuring and sample processing equipment	
Fish identification field guide	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Sampling fish communities using gill nets

Version: February 2018

11. Sampling fish communities using gill nets

11.1. Purpose and scope

This document describes the basic use of a gill net to provide a representative sample of the local fish community or to target individual species for specific purposes. This method does not collect very small fish (e.g. *Hypseleotris* spp.) and users should consider using a gill net as one component of a suite of different fishing gear types for sampling fish communities.

11.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

11.3. Health and safety

Before following the methods contained in this document, a detailed risk management (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with Queensland Work Health and Safety legislative obligations.

11.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the Fisheries Act 1994. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the Animal Care and Protection Act 2001, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes. All work carried out must comply with Australian Code for the Care and Use of Animals for Scientific Purposes (National Health and Medical Research Council 2013).

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

Note: The general fisheries permit will contain details on gill net length, drop and mesh sizes that can be used, and this must be followed. Additionally, the permit will describe how often the net must be checked and whether they can be left unattended (typically they cannot be left unattended). These instructions must be followed.

See *Permits and approvals* document for more information on requirements.

11.5. Skills, training and experience

Skills, training and/or experience required to understand and/or undertake this method include:

- prior experience in the use of gill nets
- ability to identify fish to species level.

If using a motorised boat, the driver must hold a recreational boat licence, be skilled in operation of craft while using this technique and the boat must be in survey.

11.6. Equipment

See Appendix 1 for example equipment checklist.

11.7. Procedure

11.7.1. Preparation for sampling

1. Check the specifications of the gill net to be used (i.e. length, drop and mesh size) are suitable for the aims of the sampling. The following provides examples of various gill net sizes to use for different purposes:
 - If targeting larger fish species (e.g. carp) a 150mm mesh size gill net may be the most suitable. If targeting a small to medium fish species (e.g. Hyrtl's catfish) a smaller mesh size of 25-31mm may be more suitable.
 - If sampling the fish community, use a range of mesh sizes (e.g. 25, 31, 44, 56, 75, 100, 125 and 150mm) to sample a size-range of fish.
 - Panel nets (containing randomly arranged various mesh sizes across the length of the net) are commonly used, and are useful to sample fish communities and target a range of sizes.
 - Length and drop of the net will depend upon the size of the area to be sampled and the habitat being targeted. For example, deep waters can be sampled using a gill net with a drop of up to 3m, whereas shallower waters can be sampled with a smaller drop (e.g. 1.5m). Longer gill net lengths (e.g. up to 30m) may be used in larger river systems, whereas smaller lengths (e.g. 10m) may be more suitable in creeks, streams or pools.
2. Determine if it is possible to use a boat to deploy the nets. The use of a boat is highly recommended, as deployment is extremely difficult without a boat and almost impossible in deep water. The person who holds the recreational boat licence should be dedicated to manoeuvring the boat into position. Another person should be responsible for deploying the net.
3. Check flow conditions prior to undertaking sampling—this type of sampling is not recommended in very strong flows. Before deploying nets, check each net to ensure it is free from debris and tangles.

11.7.2. Deployment of nets

1. Select a suitable location in the water body for deployment, avoiding areas with snags and rocks that may cause tangling of the net. If deploying more than one gill net, position the nets so that all nets are acting as independently as possible.
2. Tie off the upper float line of the net onto a fixed structure using a suitable knot that can be untied under load. Use additional rope where required.
3. Attach a weight to the lead line of the net before deploying in order to limit movement of the net during its deployment.
4. Begin deploying the net, while the person steering the boat slowly reverses the boat diagonally out into the current away from where the net was tied. Set the net perpendicular to the bank in still or slow-flowing water, or increasingly angled downstream with increasing flow velocity (**Figure 56**).
5. Attach a weight to the lead line before releasing the last section of the net. Attach a float to the float line to make it visible to other boaters. Include a flashing light if visibility is poor or if fishing at night.
6. Record deployment duration. The amount of time that the nets are deployed should be relatively similar for each site sampled.
7. Check the nets throughout deployment, at 30 minute or less intervals. Any captured fish should be carefully removed to minimise loss of scales and slime (a knife can be used to cut mesh if needed).



Figure 56: Setting a gill net that has been tied at one end to a snag

11.7.3. Retrieval of nets

1. Record retrieval time to assist in calculation of effort measures. If multiple nets are used, retrieve in the order they were deployed.
2. Begin to pull the net into the boat, working from the outer end of the net, removing any fish from the net as you do so.
3. Deposit fish catch in a holding container (e.g. Nally® bin) on the boat containing aerated stream water. If catch volume is small, it may be possible to process directly from the net.
4. Record details about the site such as the net measurements, number of nets used and where they were situated, habitat type and depth.

11.7.4. Catch processing

Complete processing as quickly as possible to minimise stress. See Fish holding, identification and measurement of length and weight document for further information.

11.7.5. Cleaning and maintenance of nets

Rinse the gill net after all of the fish have been released. To dry the net, tie each end to a raised structure or tree branch. Check the net often, as birds and other animals could possibly become tangled in the net. Take particular care to ensure no pest species or pathogens will be transferred between sites.

11.8. References and additional reading

Gray, CA, Jones, MV, Rotherham, D, Broadhurst, MK, Johnson, DD and Barnes, LM 2005, Utility and efficiency of multi-mesh gill nets and trammel nets for sampling assemblages and populations of estuarine fish, *Marine & Freshwater Research* 56, 1077-1088.

Marshall, J, Steward, A, McGregor, G, Marshall, C, Negus, P, Johnson, D, Lobegeiger, J, Davies, P, Harch, B, Winning, M, Choy, S, and Cockayne, B 2003, Condamine-Balonne Integrated Monitoring Pilot Project: Methods, Aquatic Ecosystems Technical Report No. 41, Department of Natural Resources and Mines, Brisbane.

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Gill nets (number and size specifications depend upon sampling design)	
Net mending kit (including extra net mesh)	
Boat, outboard motor and safety gear	
Float and light (where needed) for the river end of each net	
Anchor for the river end of each net	
Net weight for the bank end of each net	
Large holding containers (e.g. Nally® bins)	
Portable aerator (with spare batteries) with air hose and stone	
Torches, headlamps (if night work)	
Gloves for handling fish (optional)	
Fisheries permit signs	
Fish measuring and sample processing equipment	
Fish identification field guide	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Sampling fish communities using gill nets

Version: February 2018

12. Sampling fish communities using cast nets

12.1. Purpose and scope

This document describes the basic use of a cast net as part of sampling designed to either provide a representative sample of the local fish community or to target individual species for specific purposes. As use of a cast net alone may not fulfil these objectives, users should consider using a cast net as one component of a suite of different fishing gear types.

12.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping including taking field photographs and videos*

Biological assessment:

- *Background to fish sampling and index calculation*
- *Fish holding, identification and measurement of length and weight*

12.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

12.4. Permits and approvals

A general fisheries permit is required for all work that involves 'fish' as defined in the *Fisheries Act 1994*. Note that early life stages such as eggs, spat or spawn of fish are considered as fish under the Act. Under the *Animal Care and Protection Act 2001*, prior approval in writing from an Animal Ethics Committee is required for the use of animals for scientific purposes.

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park).

See *Permits and approvals* document for more information on requirements.

12.5. Skills, training and experience

Skills, training and/or experience required to understand and/or undertake this method include:

- ability to identify fish to species level
- experience in the use of cast nets.

12.6. Equipment

See Appendix 1 for example equipment checklist.

12.7. Procedure

12.7.1. Preparation for sampling

Check the cast net for any tangles or debris caught in the net. If you haven't used a cast net previously, it may be suitable to practice various techniques on a clear area of land prior to sampling to find one that suits you best.

12.7.2. Sampling

There are many techniques for throwing a cast net (e.g. off the shoulder, elbow, wrist or hand) and it will take practice to get the best technique. It will also depend on whether you prefer left or right handed throwing. The aim

is to throw the net to allow it to fully open up and catch fish. There are a variety of online resources and books with details on cast netting that can be referred to for further information. General steps to cast netting are outlined below.

1. Select a suitable section of water for cast netting that is free of woody debris, rocks, algae or plants that may snag the cast net. It may be impractical to use cast netting as a means of fishing in some streams and waterbodies due to the presence of snags.
2. Secure the hoop on the hand-line drawstring at the top of the net around your wrist.

3. With the excess drawstring, make loops and order these within the palm of the same hand to which the hoop is attached



4. Working from the top of the net, measure off one loop of net and hold it in the throwing hand (in this case, the right hand is used as the throwing hand). The length of the loop will differ depending upon length of net, height and personal preference.



5. The net should be spread out forming a rough triangle shape. With your left hand, reach down to the bottom of the net and pull up the upper section of the leadline. Place this leadline into the right hand (still holding the loops).



6. With your left hand, collect the right side of the leadline, also gathering up handfuls of this side of the net, placing it carefully into your right hand.



7. Use your left hand to pick up a leadline section of the left hand side of the net, and gather handfuls along this side of the net and hold in your left hand. The net is now ready to be cast.



8. With the majority of the net held in your right hand, extend your right arm out, parallel to the ground. Place feet approximately shoulder width apart, with the left foot forward. Extend your left arm across your body, with arms approximately shoulder-width apart, and the left arm slightly lower than the right arm. Try to keep your lower body in the same place, and let your upper body twist. The throwing movement is similar to throwing a discus.



9. To release the net, the right arm will come over to the left shoulder in a rapid movement. Follow the throw all the way through with your arms, releasing the net from both hands.



10. Once the net hits the water, it will sink to the bottom. Slowly pull the drawstring to close the net and capture fish.



11. Carefully pull in the net, quickly retrieving captured fish.

12.7.3. Catch processing

Unless the fish can be quickly processed directly from the wet net to avoid double-handling, the fish catch should be transferred to a holding container (e.g. Nally® bin) with aerated stream water. See *Fish holding, identification and measurement of length and weight* document for further information.

12.7.4. Cleaning and maintenance of nets

- Rinse, clean and dry cast net before leaving an assessment site to prevent transfer of pathogens and pest plant and animal species to other locations.
- Ensure net is maintained free of debris and holes.
- Clean/repair as necessary before storage or redeployment.

12.8. References and additional reading

Online resources with step-by-step guides to throwing a cast net include:

- <http://www.fishingnoosa.com.au/Page34.htm>

- <http://www.catfishedge.com/how-to-throw-cast-net/>
- <http://www.wikihow.com/Throw-a-Cast-Net>
- <http://www.castnetworld.com/cast-net-throwing.html>
- <http://www.gcfishing.com/castnet.html>

Appendix 1

Table 1: Equipment checklist

Equipment	✓
Cast net (available in various sizes)	
Buckets	
Large holding containers (e.g. Nally® bin)	
Portable aerator (with spare batteries) with air hose and stone	
Gloves for handling fish (optional)	
Fisheries permit signs	
Fish measuring and sample processing equipment	
Fish identification field guide	
Field data sheets	
Waterproof marker, pens and pencils	

Note: Equipment numbers/amount to be determined by the study design.

Fish collection and dissection for the purpose of chemical analysis of tissues

Version: February 2018

13. Fish collection and dissection for the purpose of chemical analysis of tissues

13.1. Purpose and scope

The purpose of this document is to provide a standard method of fish dissection to collect tissues that can be used to assess whether toxic contaminants are the cause of a fish kill. Tissues will vary between and within species (e.g. size, colour). The primary tissues to be collected include the gills, liver, and muscle (flesh). If the fish are very small, whole fish should be collected.

It is important that you have a plan in place with a suitable analytical laboratory prior to a fish kill event. Contact the analytical laboratory prior to undertaking sampling and/or dissections to determine the amount of tissue required for a particular analysis.

This document does not outline methods for assessment of disease.

13.2. Associated documents

Sampling *design and preparation*:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*
- *Choosing a laboratory and analytical method, holding times and preservation*

Biological assessment:

- *Sampling fish communities using fyke nets*
- *Sampling fish communities using bait traps*
- *Sampling fish communities using gill nets*
- *Sampling fish communities using electrofishing*
- *Sampling fish communities using seine nets*
- *Sampling fish communities using cast nets*
- *Fish holding, identification and measurement of length and weight*

13.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

13.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). Specific to this procedure, a General Fisheries Permit, Scientific user registration and animal ethics approval are required to collect live fish samples. If fish are sick or dying (i.e. during a fish kill event), the collection of fish for initial diagnosis does not require animal ethics approval. However, any further surveys outside of the initial fish kill event, such as sampling at control sites or for structured surveillance, will require animal ethics approval. See Permits and approvals document for more information on requirements.

13.5. Skills, training and experience

It is desirable that the person conducting this procedure, has some experience or training in the dissection of fish for target tissues and organs.

13.6. Equipment

See Appendix 1 for example equipment checklist.

13.7. Procedure

13.7.1. Collecting fish

Note: Taking fish or fish samples in excess of the minimum required is recommended because further investigations may not be possible if insufficient samples are taken.

- As a rule of thumb, a tissue sample of at least 20g is suggested for each analyte type (i.e. inorganics/organics). It is recommended to collect enough tissue for both organic and inorganic analyses. Providing smaller amounts of tissue sample may lead to higher limits of reporting.
 - If dissection is not possible within 24 hours, whole fish may be frozen and tissue samples taken prior to chemical analysis, although freezing may lead to rupture of internal organs.
 - If organic contaminants are to be analysed, the lipid concentration of the organ must be measured by the laboratory.
1. Determine the number of fish to be collected, and how much tissue is required from each fish for the analyses to be conducted.
 - For toxicant analysis, collect at least three fish of the same species of approximately uniform size per site if possible (to enable a statistically sound test against standards or between sites).
 - If the fish are small, you may need to collect more than three (enough tissue per sample for the lab analysis to be done). If there is insufficient quantities of tissue in each fish for analysis, then multiple fish and organ samples per site should be pooled to produce three composite samples per site.
 2. Collecting appropriate samples:
 - In order to obtain fresh tissue after a fish kill, it is preferable to choose fish that are sick or dying rather than dead (e.g. some might be moving but showing signs of lethargy or distress). When sampling live fish, ensure that the fish are handled and euthanized humanely (such as with the use of AQUI-S™¹⁶). If only dead fish are present, choose the least decomposed fish available.
 - If collecting fish for chemical analysis other than in response to a fish kill, follow the relevant fish sampling procedures listed in the Biological assessments documents within this manual.
 3. Place individual samples into individual resealable bags with a label stating relevant information such as date, time, sampler details, site, species and replicate number.
 4. Place samples in an esky with crushed ice to transport to a laboratory, or clean area if dissections are to be carried out in the field.

13.7.2. Preparing for dissection

1. Ensure there is a clean working area and that equipment can be rinsed between each sample.
2. Clean sampling equipment to be used for the dissection:
 - Tools, work surface, and sample containers¹⁷ must be clean and not likely to contaminate the samples with an analyte of interest (for example, if nickel or chromium are of interest, then stainless steel tools may be inappropriate).
 - After each fish is dissected, all equipment should be cleaned and rinsed, and the cutting board covering and gloves need to be changed.
3. Clear an area to conduct the dissection.
4. Place clean aluminium foil (for organics/pesticides analysis) or plastic (for metal analysis or other) on the cutting board or tray prior to placing the fish on the work area.
5. Set up the work area to ensure all equipment is easily accessible once dissections begin.
6. Place a waste bin in an area easily accessible to the person conducting the dissections.
7. Identify a procedure for naming each sample/replicate/organ, and relating these back to the individual sample. See the Preparation for sampling document for information regarding naming sites and samples.

¹⁶ Other acceptable euthanasia methods are described in ANZCCART, 2001, Euthanasia of Animals Used for Scientific Purposes Reilly (2nd edition), Department of Environmental Biology, Adelaide University. Available from: <https://www.adelaide.edu.au/ANZCCART/docs/euthanasia.pdf>

¹⁷ If high concentrations of contaminants are suspected, laboratory grade plastic bags should be adequate to store tissues in. However, if low concentrations of an analyte are suspected it may be more appropriate to store the fish organs in glass jars that have been supplied and cleaned specifically for the analyte of interest by the analytical laboratory.

13.7.3. Fish dissection

1. Measure and weigh fish in accordance with the Fish holding, identification and measurement of length and weight document. Record details.
2. Put on powder-free gloves. Gloves must be stored in a clean environment (e.g. in a resealable plastic bag).
3. Lay fish flat on one side with the dorsal fin facing away from you.

13.7.3.1. Gill samples

If the gills are to be collected:

1. Lift the operculum (gill cover) (**Figure 57**) and cut this off at its base to expose the gills (**Figure 58**). Take care not to damage the gills when doing this.
2. Carefully cut out the gills at their base (**Figure 59**), taking care not to damage these when doing so.
3. Rinse gills with de-ionised water.
4. Place gills in labelled storage container/bag (see Section 13.7.4).

Note: Gills on larger fish may not require the operculum to be removed.



Figure 57: Lifting the operculum (gill cover)



Figure 58: Cut at the base



Figure 59: Cut gills at the base

13.7.3.2. Muscle samples

Muscle (flesh) samples should be collected above the lateral line, between the dorsal fin and the caudal fin. This will maximise the amount of muscle tissue collected and reduce the risk of accidentally piercing internal organs. Avoid cutting below the fish's lateral line to ensure the lower intestine or other internal organs are not pierced. If the intestine is cut open, this will lead to contamination of the organs and the sample will not be usable.

If muscle is to be collected:

1. Make a cut with the scalpel blade from just below the start of the dorsal fin down to the fish's lateral line (Cut 1, **Figure 60**).
2. Cut from just above the lateral line of the fish toward the tail (Cut 2, **Figure 60**).
3. Cut from where the first incision was made just below the dorsal fin across the top of the fish and down toward the tail (Cut 3, **Figure 60**), to meet the cut from step 2.
4. Remove the skin of this section of cut flesh using forceps and a scalpel blade (**Figure 61**). Take care not to touch this exposed muscle.
5. To remove the muscle sample, make incisions around the dissected area, cutting underneath the flesh to detach it from the small bones and allow it to be removed (**Figure 62**).
6. Once the muscle has been removed from the fish, rinse it in deionised water.
7. Place muscle sample in labelled storage container/bag (see Section 13.7.4).

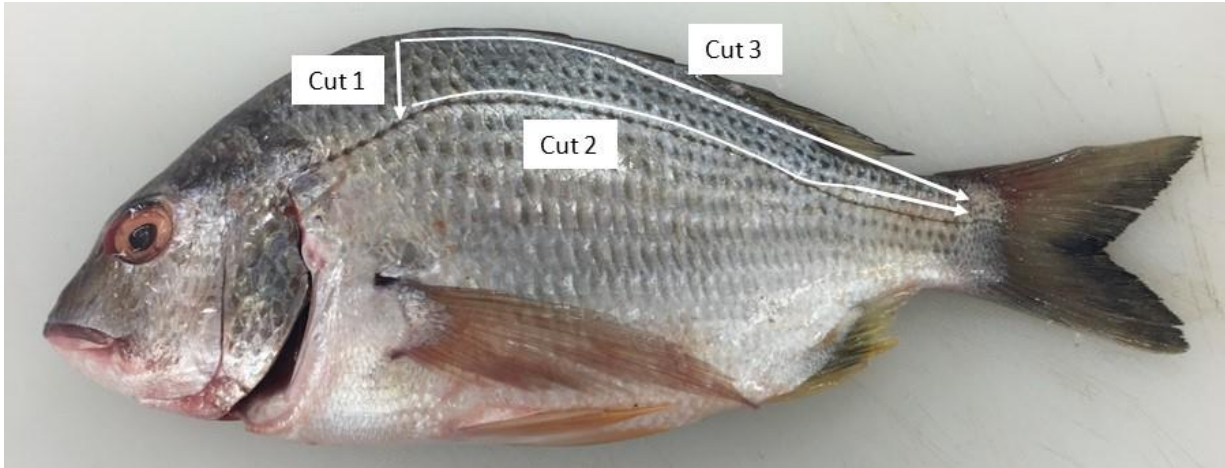


Figure 60: Outline of area to be removed from the fish for muscle sample



Figure 61: Removing skin



Figure 62: Removing muscle

13.7.3.3. Other internal organ samples

If other internal organs (i.e. primarily liver) are to be collected:

1. Make a small cut just in front of the anus (vent) to open the abdominal cavity.
2. With blunt-ended scissors, cut along the belly (ventral midline) of the fish, forward to the middle of the lower jaw (**Figure 63**).
3. Remove the flap of skin covering the abdominal cavity by cutting from the small cut in front of the anus upwards, across the body of the fish and toward the head of the fish (**Figure 64**). This should expose the heart and abdominal organs for examination and removal.
4. Carefully cut out the organ for examination, taking care not to damage these when doing so.

Note: Organs can be located in differing/varying places depending upon the body shape of the species (e.g. **Figure 65** and **Figure 66**). The kidney is a relatively difficult organ to locate and dissect successfully. It is usually located up close to the spine and may be hidden by the swim bladder.

5. Rinse the removed organ with de-ionised water.
6. Place the removed organ in labelled storage container/bag (see Section 13.7.4).



Figure 63: Cut along the belly



Figure 64: Expose the abdominal cavity

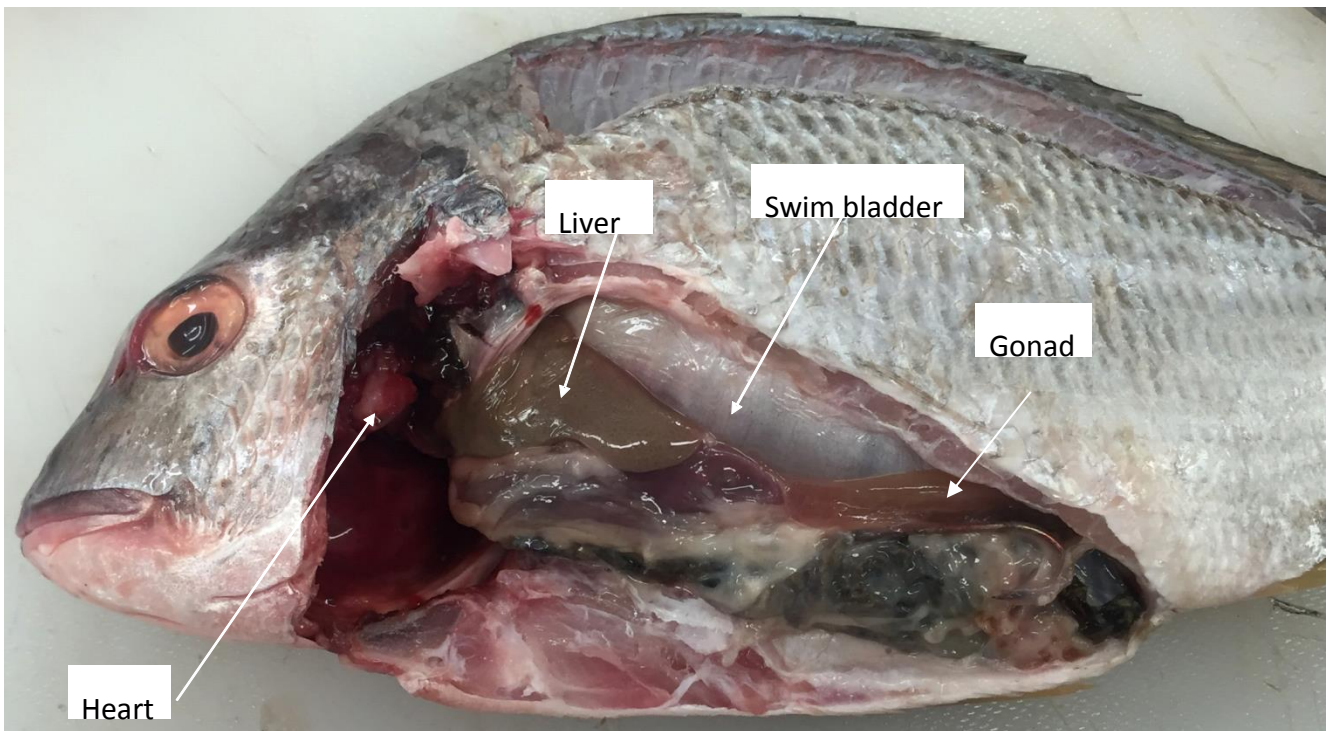


Figure 65 Internal anatomy of a yellowfin bream (*Acanthopagrus australis*)

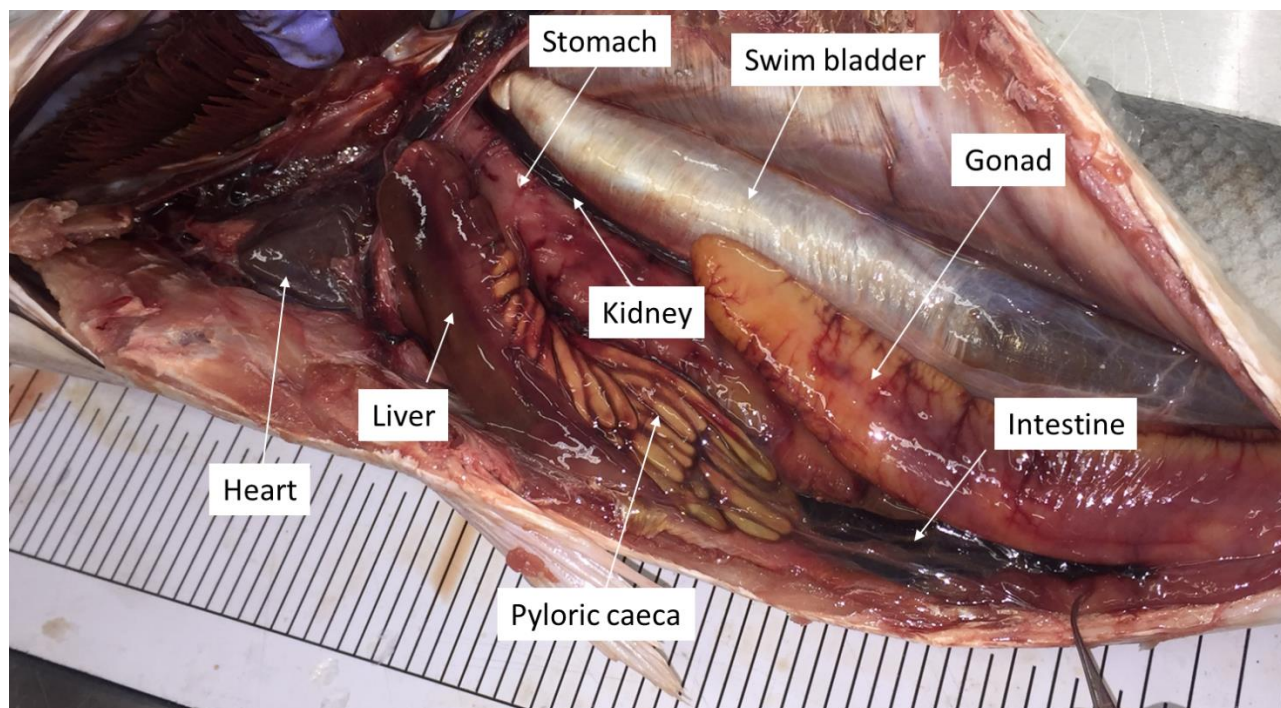


Figure 66 Internal anatomy of a tailor (*Pomatomus saltatrix*)

13.7.4. Preserving and packing samples

Packing samples will depend upon the analysis required and should be discussed with the analytical laboratory prior to dissections. Individual organs should be separately packaged, labelled and preserved prior to sending to the laboratory.

For toxicant analysis:

- where poisoning by pesticides or organic compounds is suspected:
 - wrap fish or samples in aluminium foil with the dull side of the foil inwards
 - place in laboratory grade container or bag
 - freeze as soon as possible (-20°C freezer for longer term storage i.e. >24–48h)
- for other analysis, including metals
 - place fish or samples in laboratory container or bag
 - freeze as soon as possible (-20°C freezer for longer term storage i.e. >24–48h)

13.8. References and additional reading

There is a vast array of online resources with information on fish anatomy for various species of fish, and step-by-step guides to fish dissections. These guides and videos will assist with fish dissections. Some examples include:

European Association of Fish Pathologist (a highly comprehensive manual for fish necropsy)

<http://necropsymanual.net/en/>

<http://www.popstoolkit.com/sops/methods/fish.aspx>

<http://www.jove.com/video/1717/dissection-of-organs-from-the-adult-zebrafish>

USEPA(United States Environment Protection Agency), 2000. Guidance for Assessing Chemical Contaminant Data for Use in Fish Advisories. Volume 1. Fish Sampling and Analysis. Third Edition. Available from: <https://www.epa.gov/sites/production/files/2015-06/documents/volume1.pdf>

USGS (United States Geological Survey), Biomonitoring of Environmental Status and Trends (BEST), Filed Procedures for Assessing the Exposure of Fish to Environmental Contaminants. Available from: <https://pubs.er.usgs.gov/publication/itr19990007>

ANZCCART(Australian & New Zealand Council for the Care of Animals in Research and Teaching) 2001, Euthanasia of Animals Used for Scientific Purposes Reilly (2nd edition), Department of Environmental Biology, Adelaide University. Available from: <https://www.adelaide.edu.au/ANZCCART/docs/euthanasia.pdf>

Appendix 1

Table 1 Equipment checklist

Equipment	✓
Various sizes of laboratory grade resealable plastic bags (if analysing for inorganics/metals), and/or laboratory supplied and cleaned glass jars for the analyte of interest	
Aluminium foil (if analysing for organics/pesticides), or clean plastic sheet (metals or other)	
Scalpel (with disposal blades and sharps disposal unit)	
Scissors	
Forceps	
Disposable gloves (powder-free)	
Marker pen for labelling samples	
Measuring board and scales (to measure/weigh fish prior to dissection)	
Tray or cutting board to dissect upon (if possible)	
Table	
Waste bucket	
De-ionised water	
Squeeze bottles	
Portable fridge/freezer (-20°C freezer for longer term storage i.e. >24-48h)	
Fish catching equipment and associated euthanasia equipment	

Background to aquatic macrophytes, collecting data along a belt transect

Version: February 2018

14. Background to aquatic macrophytes, collecting data along a belt transect

14.1. Purpose and scope

This document provides background information on aquatic macrophytes and collecting data along a belt transect.

14.2. Associated documents

Biological assessment: Aquatic macrophytes, collecting data along a belt transect

14.3. Introduction

Aquatic plants (also known as macrophytes) are defined as plants 'that grow in water or need a waterlogged environment to carry out their life cycle'¹⁸. An aquatic macrophyte can be an emergent as well as being submerged or floating (**Figure 67** and **Figure 70**).

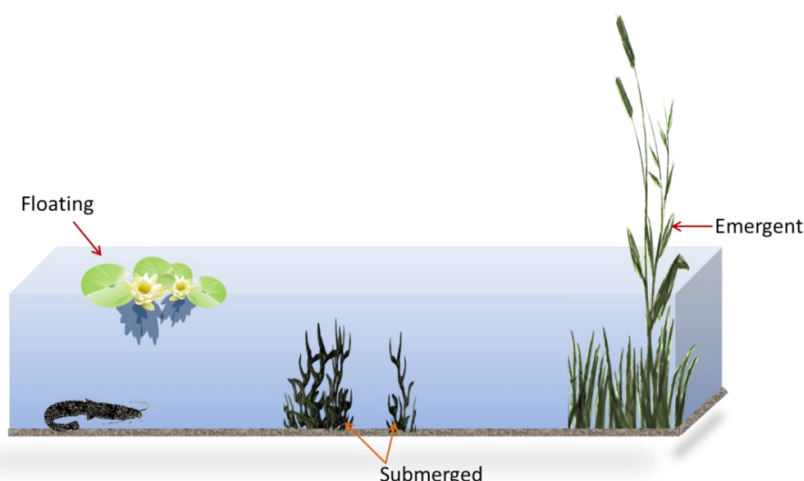


Figure 67: Schematic showing submerged, floating and emergent vegetation

Because they are supported by the water, aquatic macrophytes need little of the supporting or structural tissues that are found in land plants. Instead, there are numerous air spaces inside the stems, leaves and roots that aid gas exchange between the shoot and the root and also aid buoyancy. Submerged parts generally have no, or alternatively, only a thin waxy cuticle enabling the plants to absorb minerals and gases directly from the water.

Aquatic macrophyte habitats can occur in slow to fast flowing water. In lakes (**Figure 69a**) and rivers these plants are important because they provide cover for fish, water birds and a solid substrate for aquatic invertebrates. They also produce oxygen, which aerates the water, and are an important food source for some fish, birds and other wildlife.

¹⁸ <http://wetlandinfo.ehp.qld.gov.au/wetlands/ecology/components/flora/>

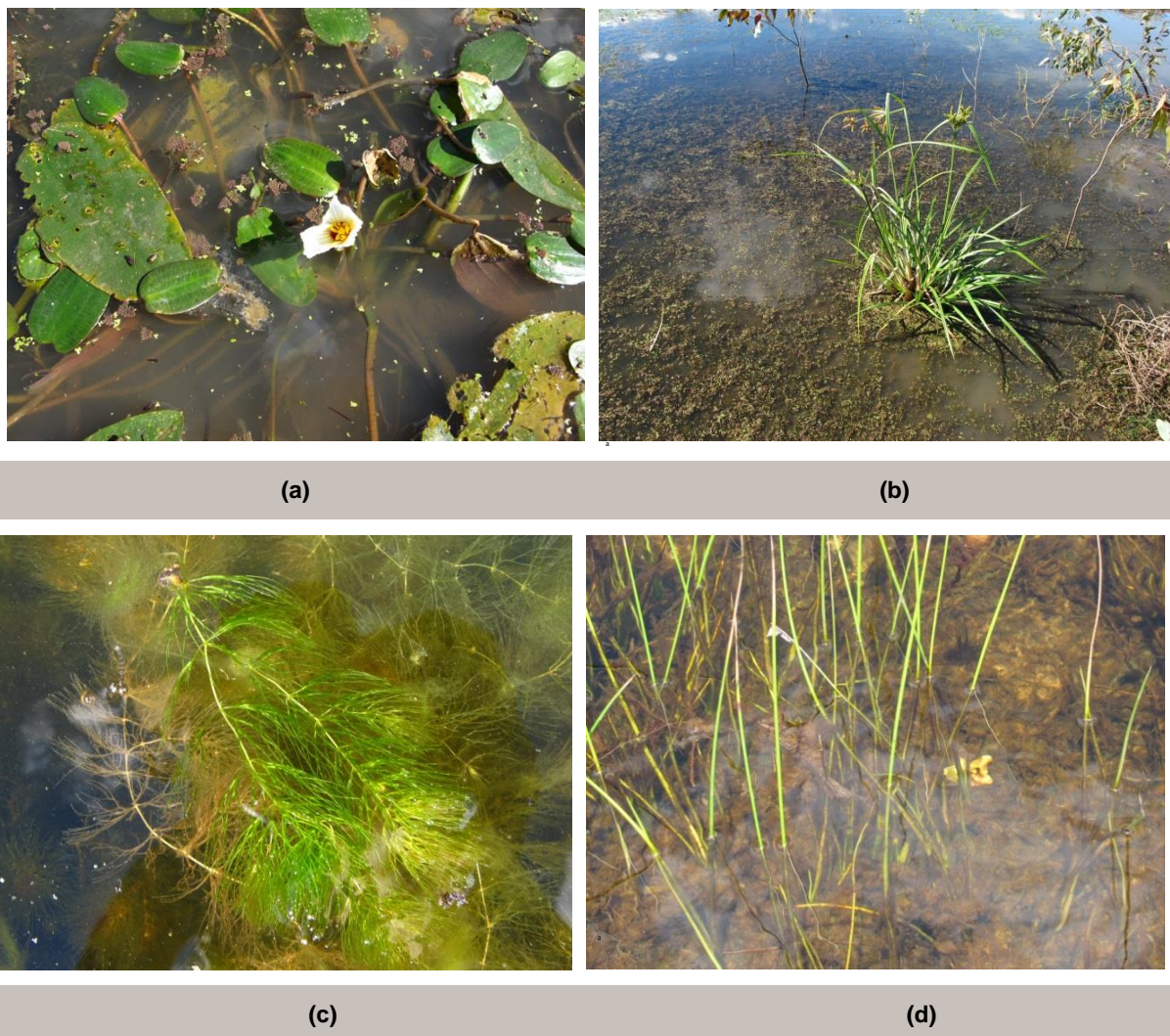


Figure 68: Native aquatic macrophytes (a) floating (*Ottelia* sp.), (b) emergent (*Cyperus* sp.), (c) submerged/free floating (*Ceratophyllum* sp.) (d) emergent (*Eleocharis* sp.)

14.4. Macrophytes as environmental indicators

Aquatic plant species assemblages are often determined by environmental variables at the local and landscape level. Macrophyte composition, abundance and growth are useful environmental indicators because they can be affected by a number of physical and chemical factors within stream habitats, including turbidity, nutrient concentrations and flow disturbance regimes. Macrophytes are not only affected by environmental conditions, but they themselves facilitate changes in water chemistry and physical habitats and can have a major role in aquatic ecosystem functioning, including:

- provision of habitat for aquatic organisms such as macroinvertebrates and fish
- reduction of erosion on stream banks
- effects on the nutrient cycle
- vertical mixing of water
- increase in dissolved oxygen levels
- reduction in water velocities, increase in water depth and channel width
- increase in sedimentation
- act as a food source.

A decline in a macrophyte community recorded over time may indicate water quality problems and changes in the ecological status of the water body. Land use practices can cause numerous impacts on water quality which can in turn impact on macrophyte communities via excessive turbidity and sedimentation, (deposition of mineral or organic matter by a fluid flow) as well as increases in nutrient concentration and herbicides.

High turbidity can result in a low abundance and low species diversity of submerged aquatic plants. However, in Australia, shallow lakes and reservoirs may have high natural turbidity because wind results in the resuspension of sediments, and can represent a healthy natural habitat. Also, lakes and reservoirs in catchments with highly

dispersible soils will have naturally high turbidity (ANZECC and ARMCANZ, 2000).

Increased nutrient concentrations, can lead to an over-abundance of floating macrophytes (**Figure 69b**). These include native species such as *Azolla* spp. (**Figure 69c**) and in the number of invasive weeds such as *Salvinia molesta* (**Figure 69d**). *Salvinia molesta* is a particularly harmful weed in waterways around Australia because excessive growth of this plant can choke streams and rivers causing serious environmental damage, including the loss of native species.

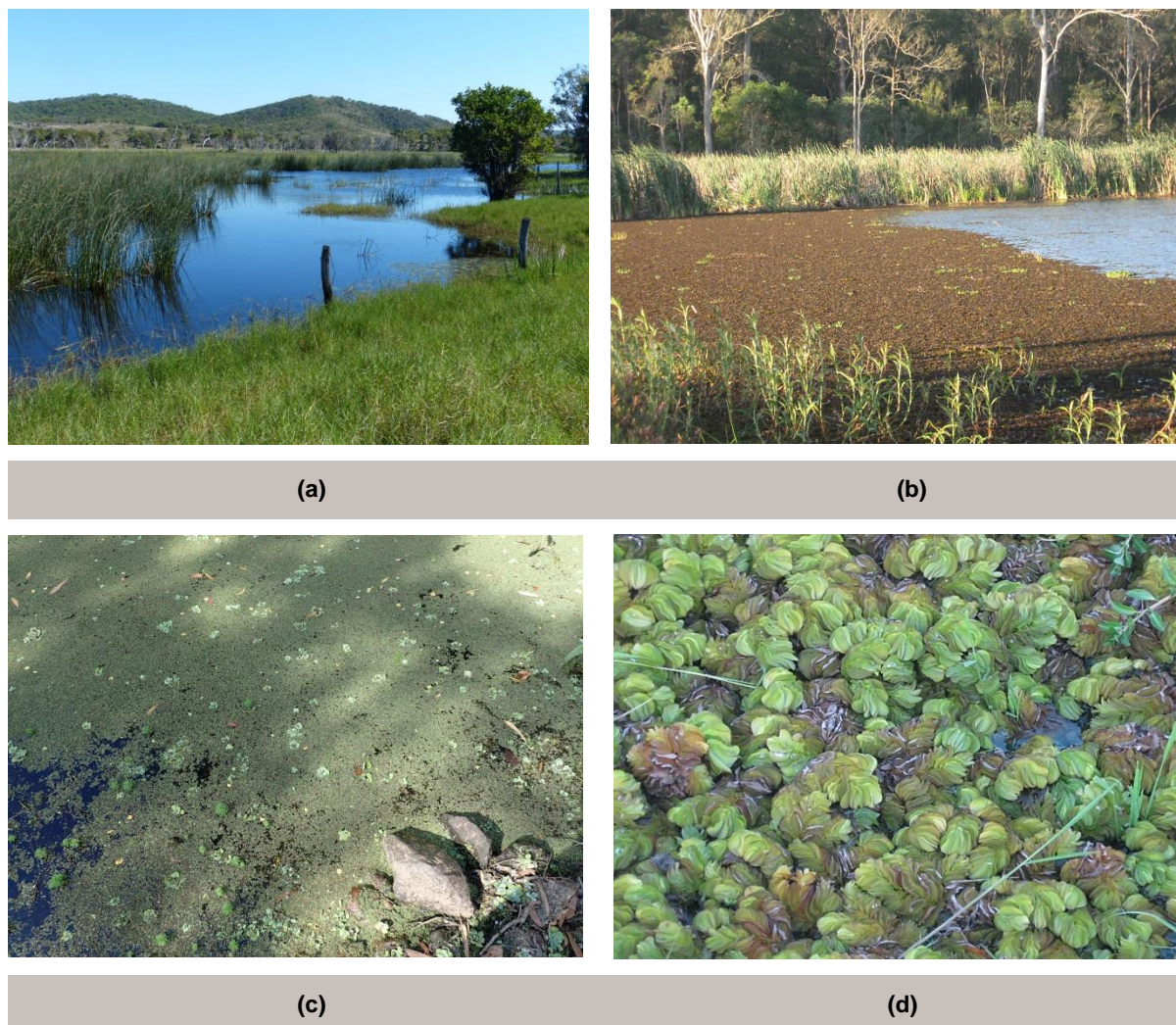


Figure 69: (a) Healthy aquatic macrophyte lake habitat (b) impacted lake habitat with abundant growth of invasive floating aquatic weed and native emergent species (c) native aquatic fern, *Azolla* sp. and (d) introduced weed, *Salvinia molesta*

Aquatic macrophytes can be monitored by collecting data along a belt transect as described in Aquatic macrophytes, collecting data along a belt transect document.

14.5. References and additional reading

ANZECC and ARMCANZ 2000, Australian and New Zealand Guidelines for Fresh and Marine Water Quality, National Water Quality Management Strategy, Paper 4, Australian and New Zealand Environment and Conservation Council and Agriculture and Resource Management Council of Australia and New Zealand, Canberra.

Barrat-Segretain, M-H 2001, 'Biomass allocation in three macrophyte species in relation to the disturbance level of their habitat', *Freshwater Biology* 46, 935-945.

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Aquatic macrophytes, collecting data along a belt transect

Version: May 2018

15. Aquatic macrophytes, collecting data along a belt transect

15.1. Purpose and scope

Macrophyte data can be collected by establishing belt transects and/or quadrats at a site. This document describes how to collect aquatic macrophyte data in the field using a belt transect.

This method is difficult to employ in water that is highly turbid. Additionally, the method may not be appropriate for collecting macrophyte data from deep sections of water, although the likelihood of macrophytes growing in deep sections may be low.

15.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological assessment: Background to aquatic macrophytes, collecting samples along a belt transect

15.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

15.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

15.5. Skills, training and experience

At least one staff member must have experience in identifying aquatic macrophytes.

15.6. Equipment

See Appendix 1 for example equipment checklist.

15.7. Procedure

15.7.1. Locating the belt transect

A belt transect is a defined area, usually a rectangle, of specified size within which aquatic macrophyte data can be recorded. The dimensions of the belt transect are defined by the objectives of the monitoring program. The boundary of the belt transect can be defined with a tape measure, or visually using a range finder. The edges of the river can define two of the edges of the belt transect.

Data can be collected from:

- a location where a specific habitat is present
- a number of belt transect positions used to represent a site. These may encompass a number of different flow, substrate and depth combinations. For example, four 10m belt transects could be established across the channel width within a 100m reach of stream, with each belt transect separated by 20m (**Figure 70**). The number of belt transects used and the distances between each will depend upon the monitoring objectives.

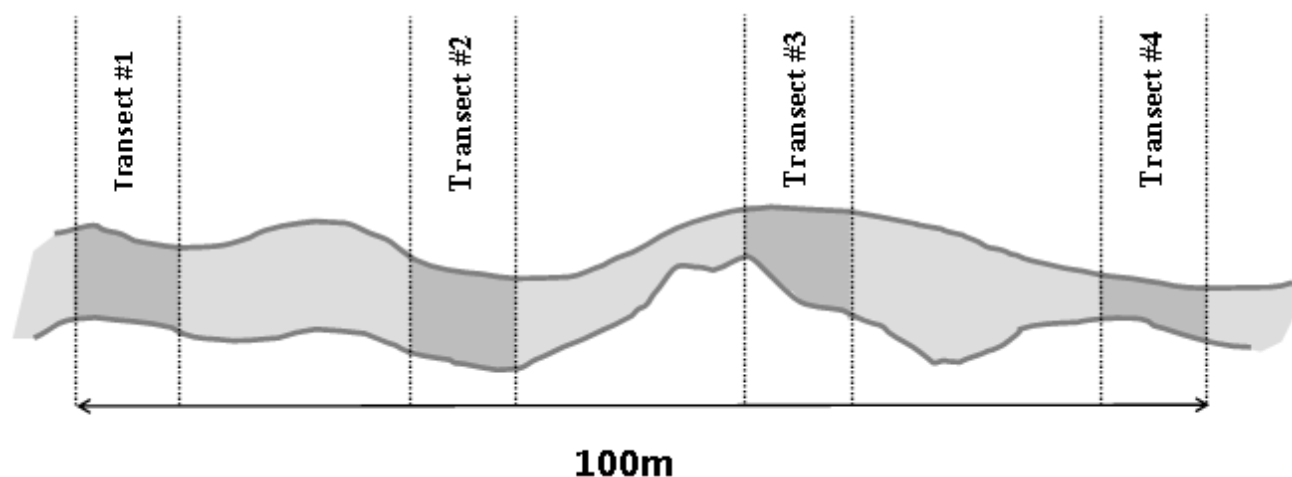


Figure 70: Example of a belt transect placement

15.7.2. Data collection

Macrophyte presence and relative abundance should be estimated along each transect. Macrophyte relative abundance can be recorded as the per cent cover of plant material per species above the substrate when viewed from above. The per cent cover for each taxon can be scored using either numerical (Table 9) or descriptive (Table 10) notation. The total per cent cover for all aquatic macrophyte taxa in a belt transect can equal more than 100 per cent, because macrophyte species from different growth forms can occur in the same location but at different depths. For example, floating and submerged macrophyte species can occur at the same position along the belt transect. In order to collect the data along a transect:

- 1 Position the belt transect within the sampling reach using a tape measure or rangefinder.
- 2 Work from the most downstream belt transect to the most upstream belt transect in order to avoid obscuring the view into the water by stirring up sediment and debris.
- 3 Wade into the water to the location of the belt transect.
- 4 Wade along the length of the belt transect, looking down into the water from above at the macrophytes present.
- 5 At the end of the belt transect record the required data (e.g. macrophyte species presence and relative abundance) onto a field sheet (Appendix 2).
- 6 Repeat the process for other belt transects.

Table 9: Macrophyte cover as a numerical score

Category	Per cent cover of macrophyte taxon
1	0%
2	1–5%
3	6–25%
4	26–50%
5	51–75%
6	76–95%
7	96–100%

Table 10: Macrophyte cover as a descriptive category

Category	Per cent cover of macrophyte taxon
Absent (A)	0%
Isolated (I)	1–5%
Scattered (S)	6–20%
Beds/Stands (B)	21–50%
Overgrowing/Filling Channel (O)	51–100%

15.8. Confirmation of species identification

Representative samples of aquatic macrophytes that cannot be identified in the field should be collected and pressed¹⁹ for later identification using appropriate keys. Plants should be identified to species level where possible, and a reference herbarium can be maintained. Where a specimen cannot be identified to the species level, it should be lodged with the Queensland Herbarium for formal identification. Specimen collection and vouchering should be done in accordance with the Queensland Herbarium procedures¹⁹.

¹⁹ Queensland Herbarium (2013) Collection and preserving plant specimens, a manual. Department of Science, Information Technology, Innovation and the Arts. <https://www.qld.gov.au/environment/assets/documents/plants-animals/herbarium/collecting-manual.pdf>

15.9. References and additional reading

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Appendix 1**Table 1: Equipment checklist**

Equipment	✓
Tape measure or range finder.	
Plant press and paper.	
Aquatic macrophyte identification field guides	
Field sheets	
Camera, charger	
GPS	
Spare batteries	
Waders	
First aid kit	

Background to monitoring mangrove forest health

Version: February 2018

16. Background to monitoring mangrove forest health

16.1. Purpose and scope

This document provides background information on the measures of mangrove forest health.

16.2. Associated documents

Biological assessment: Monitoring mangrove forest health

16.3. Introduction

Mangroves comprise several species of trees and shrubs that grow along sheltered intertidal shorelines, predominantly in tropical and subtropical coastal environments. Mangroves are adapted to salt-water environments and grow in muds, silts, sand and anoxic marine sediments.

Mangrove forests commonly consist of several shoreward zones including *Rhizophora* (red mangrove) and *Avicennia* (grey mangrove) species (**Figure 71**). A number of mangrove species produce seeds which germinate whilst they are still attached to the tree (vivipary). In some species these seedlings may reach over 30cm in length before they separate from the parent plant and fall to the mud below or get carried by currents to root elsewhere. In the unstable coastal environments that they normally occur in, un-germinated seeds would be at a greater risk of being washed away if they had first to germinate on the ground and then anchor themselves to the substrate.

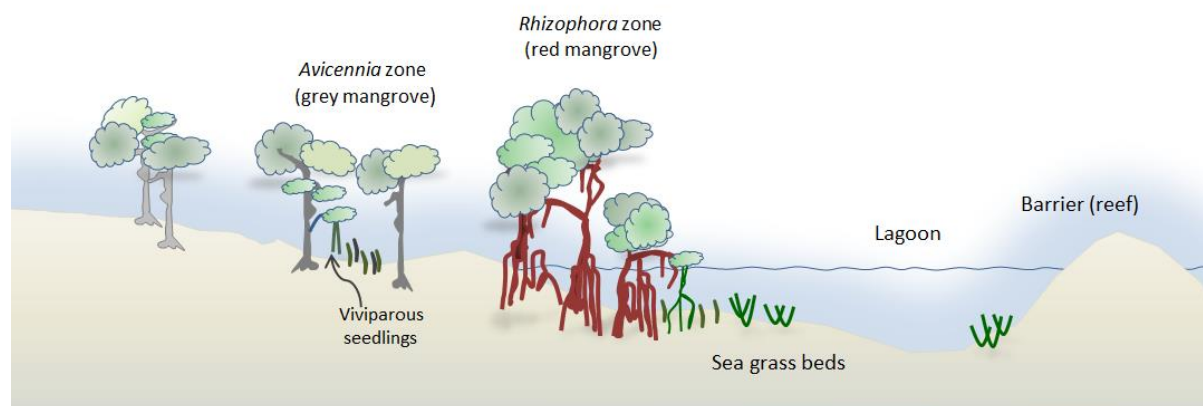


Figure 71: Schematic of mangrove zones

Dense mangrove forests located on shorelines protect the coastline from the impacts of storm waves and coastal erosion. They trap and bind sediments, capture effluents from terrestrial runoff and provide a buffer for nutrients, metals and other toxicants entering coastal waters, reducing coastal turbidity and improving water clarity.

Mangrove forest canopies and their extensive root systems are also a major source of primary productivity by providing a food source and habitats supporting both marine and terrestrial animals including fish, birds, invertebrates and insects. They are of particular economic importance, because they provide nursery habitats for commercially important fish and prawn stocks thus replenishing estuarine and coastal fisheries.

Monitoring changes to mangrove forest communities and mangrove forest health can help diagnose causes of environmental stress. Mangrove forests provide a means to monitor changes in coastal environments such as storm effects, sea level change, pollution, and sedimentation rates.

General guidance on the methods used to monitor mangrove forest health in Queensland are given below. However, it is recommended that expert advice be sought when designing a mangrove monitoring program.

16.4. Measures of mangrove health

16.4.1. Litter

Mangrove litter production is the shedding of vegetative and reproductive structures of mangroves (e.g. leaves and seeds). This may be caused by natural growth cycles, age, stress and/or mechanical factors, such as wind. This litter fall, which is part of the net primary productivity of a mangrove system, is the basis of detritus food chains.

The rate of litter production can indicate the health of a mangrove community. A healthy system will produce a stable monthly and/or yearly volume of litter as older leaves are shed and replaced with new ones. Declining production over time may indicate that a community is under stress.

The ratio of fallen leaves to stipules of *Rhizophora* species can also be used as an indicator of system health. Thus, in a healthy *Rhizophora* community, the ratio of leaves to stipules in the litter should be close to 1:1. If there are more leaves than stipules then this indicates that the plant is shedding leaves due to stress.

The main reasons for monitoring litter productivity are to:

- gain an understanding of the baseline litter productivity of a mangrove community
- indicate system health—as mangrove communities become under stress they are likely to be less productive, resulting in less litter production over time or alternatively, communities under stress produce a large amount of litter over a short period of time as the plants shed leaves
- look at unseasonably low litter production due to poor growth resulting in less detritus which may affect faunal communities.

16.4.2. Seedling regeneration

Seedling regeneration within a mangrove stand can lead to long term changes within mangrove communities. In addition seedling regeneration can give an indication of mangrove community health. The rate of growth of seedlings is a more important indicator than the number of seedlings present in a given area in determining likely long term community maintenance or change.

Long-term hydrological changes can result in sediment deposition or greater tidal and/or freshwater influence. This may result in colonisation by different species of mangroves from those that were originally present. For example, long-term sediment deposition may raise the elevation of a site, resulting in conditions more favourable for species normally found higher up the tidal gradient. Other hydrological changes, resulting in more tidal or freshwater influence, could create conditions favouring species normally found closer to the seaward or landward margin of the mangroves. Identifying what species of mangrove is replacing the previous forest can indicate whether climatic or hydrological changes have or are occurring.

16.4.3. Canopy cover and leaf area index

Leaf area index (LAI) provides an estimate of canopy cover. Since plants under stress tend to shed leaves, a reduction in canopy cover over time may indicate ecosystem stress or disturbance.

LAI is an estimate of the total area of leaf surface within a plant community relative to the ground area of that community. To calculate LAI, the intensity of full sunlight is measured (using a light meter), and this is compared with the light intensity measured under a mangrove canopy. LAI can be used to monitor short to long-term foliage patterns and changes in a mangrove stand (e.g. high rates of primary productivity during good seasons, or defoliation through storm damage, seasonal or drought-related leaf fall, insect attack, etc.).

16.4.4. Mangrove forest structure

Mangrove structure refers to the composition of a mangrove community in terms of canopy height, stem density, age, tree diameter and species present. It varies considerably between different forest types, and between the same forest types in different locations. Structure is also influenced by many natural factors including climate, tidal inundation, soil pH and salinity, sediment particle size and amount of freshwater the community is exposed to.

Mangrove structure is likely to be affected when any of these parameters are altered by human-induced impacts. Positive changes can result in greater forest vigour (increased diameter, canopy cover and stem density), while negative changes can stress the mangrove community, resulting in reduced canopy cover and stem density, tree mortality and, eventually, reduced basal area of trees and/or lower canopy height.

Higher proportions of dead versus live stems, and/or decline in the basal area of the trunks of mangroves in a stand may indicate stress or disturbance. Many aspects of mangrove structure tend to respond more slowly than other estuarine indicators and it can take several years before changes can be detected.

16.4.5. Crab burrow counts

Estuarine crabs break down much of the leaf and other organic matter produced by mangrove forests. Their burrows also increase the ratio of soil surface area to air, resulting in aeration and oxidation of the mostly anoxic mangrove soils. This oxidation can be important for the growth of mangrove plants. Consequently, changes to the crab population can affect the nutrient cycling and oxidation of intertidal soils, which in turn can affect the productivity of mangroves. Decreased crab populations and associated burrow density can lead to decreased nutrient cycling and soil aeration, and reduced production of surrounding plants.

Crabs can be sensitive to pollution. Their absence from a mangrove forest may indicate that the site is experiencing human-induced stress. Data on crab burrow density may complement leaf litter trapping or other mangrove monitoring programs.

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Monitoring mangrove forest health

Version: May 2018

17. Monitoring mangrove forest health

17.1. Purpose and scope

The scope of this document is to provide general guidance on a method that can be used to monitor mangrove forest health in Queensland. It is based on generic plot-based vegetation survey (Neldner et al. 2012) and vegetation condition assessment (Eyre et al. 2015) methods used in Queensland with additional measures e.g. litter trapping and crab burrow counting, which provide some indices on forest productivity. Alternative plotless methods may also be used, provided they are consistently applied and record the structural, floristic and productivity attributes of mangroves and evaluate change over time in these attributes.

In the case of harm to a mangrove ecosystem, it is recommended advice be sought from a mangrove expert.

17.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological assessment: Background to monitoring mangrove forest health

17.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

17.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See *Permits and approvals* for more information on requirements.

17.5. Skills, training and experience

Skills, training and or experience required to understand and/or undertake this method include the ability to identify the different species of mangroves and other plants found in mangrove forests.

17.6. Equipment

See Appendix 1 for example equipment checklist.

17.7. Procedure

All the methods outlined in this procedure should ideally be used to fully assess the health of a mangrove forest, although a subset of methods may be used based on the objective of the study or investigation being undertaken. Monitoring using all methods should, where possible, be undertaken in a single 50 x 10m plot (apart from the mangrove forest structure procedure where a number of 50 x 10m quadrats are surveyed). The alignment of this plot will depend on the width of the community being monitored but it is best to align the plot at right angles to the seaward edge of the mangroves if this is possible (**Figure 72**). Permanent markers, such as surveying pickets, should be used to identify the corners of the quadrat.

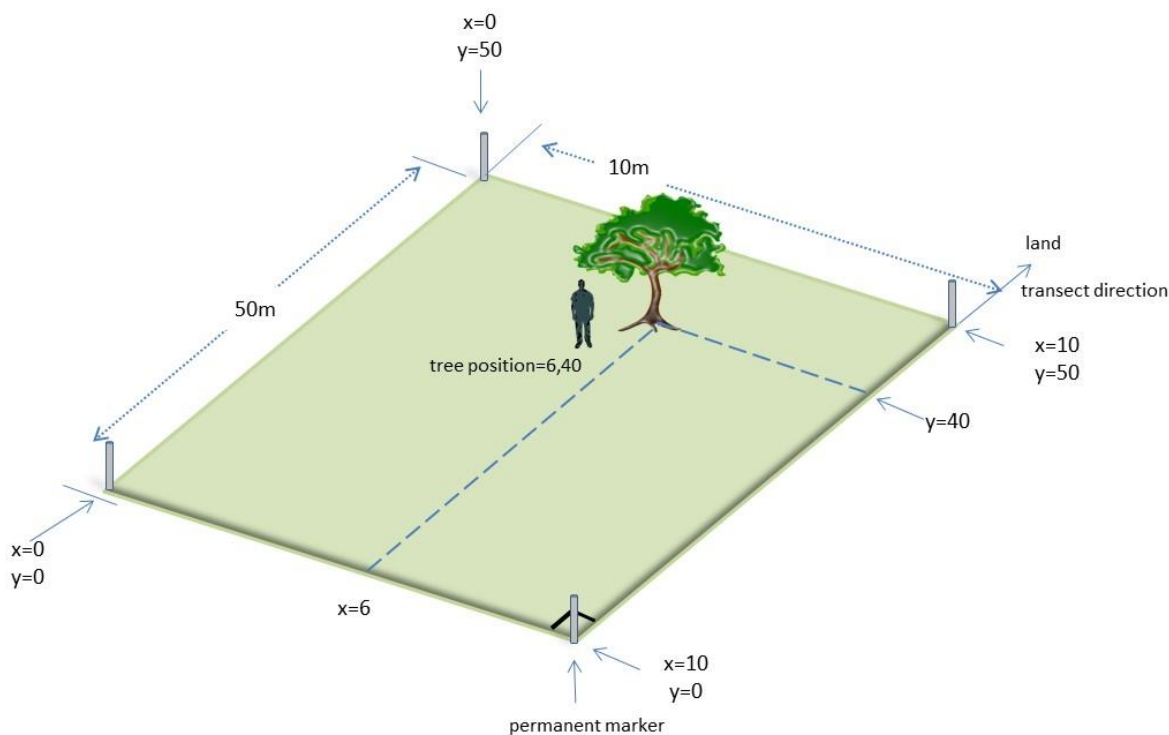


Figure 72: Setup of main sampling quadrat

17.7.1. Site selection

Though site selection will depend on the objectives of the monitoring program, in most instances it will be necessary to select sites that are representative of the mangroves in the area. Use recent aerial photographs to determine the size and extent of the site, and look for zonation patterns between the seaward and landward margins. Sites should be ground-truthed to confirm zonation patterns, and to ensure that they are representative. Select potential monitoring sites so they are representative of the mangrove community which you are trying to monitor.

As mangrove systems are diverse and can vary considerably in structure and floristics over short distances, a person of suitable experience should assist with this process.

Also note any evidence of unusual occurrences, such as deposition of rubbish, or other human-induced disturbances.

17.7.2. Mangrove litter trapping

Leaf litter traps are installed in a mangrove community and litter is collected monthly, sorted into different categories (leaves, twig, bark, flowers and propagules), oven dried, and weighed.

The dry weight of the litter is a measure of the productivity of the mangrove community.

Problems to be aware of include:

- significant disturbances (e.g. cyclones and storms) may damage sites and make it necessary to begin collecting data again for a time series
- traps can be interfered with
- climatic and biological differences in Australian mangroves make it difficult to compare data from different locations.

Within the forest:

1. Set up a transect running parallel to the tidal gradient (see **Figure 73**).
2. Put litter traps at 5m intervals along the centre line of the transect.
3. Install litter traps by attaching a nylon cord to each corner of the trap and hanging them evenly from mangrove branches (instructions for making a litter trap are in Appendix 2). Ensure that the bottom of the trap or chute is above the high tide mark (**Figure 74**).

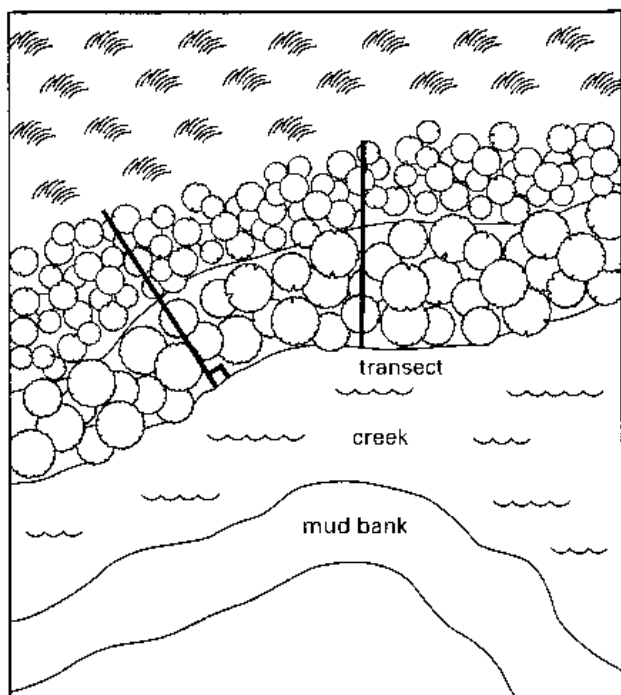


Figure 73: How to establish a transect

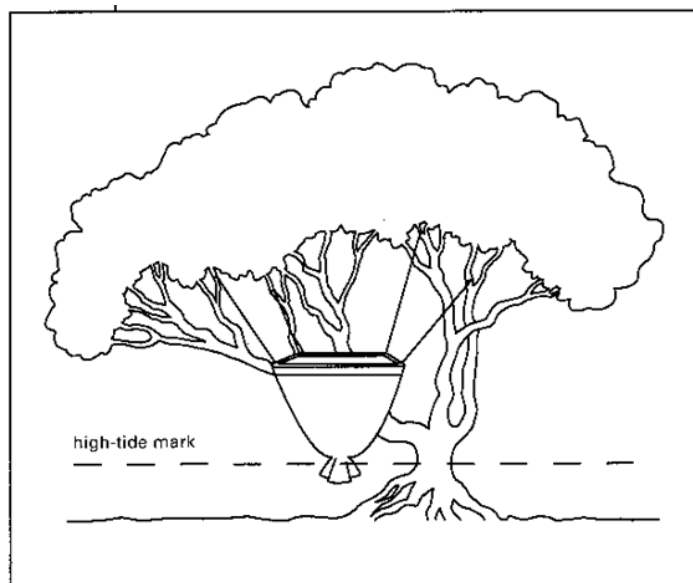


Figure 74: A litter trap in a mangrove forest

1. To empty a trap:

- 1.1. Remove any large sticks and put them in a plastic bag labelled with the trap number.
- 1.2. Put the bag under the chute.
- 1.3. Untie the chute and empty the trap contents.
- 1.4. Re-tie the chute securely.
- 1.5. Proceed to the next trap.

Note: Traps should be emptied every month (every two weeks if measuring *Avicennia* spp.) to prevent leaf decay and to determine monthly trends.

On returning from the field:

2. Sort contents from each trap (do not mix content from different traps) into leaves, flowers, bark (include wood), seeds, stipules (if monitoring in a *Rhizophora* forest) and other items if they are present.
3. Count the number of leaves and stipules and record the result.
4. Place the sorted contents into smaller, labelled paper bags, and then put the datasheet and smaller bags in the

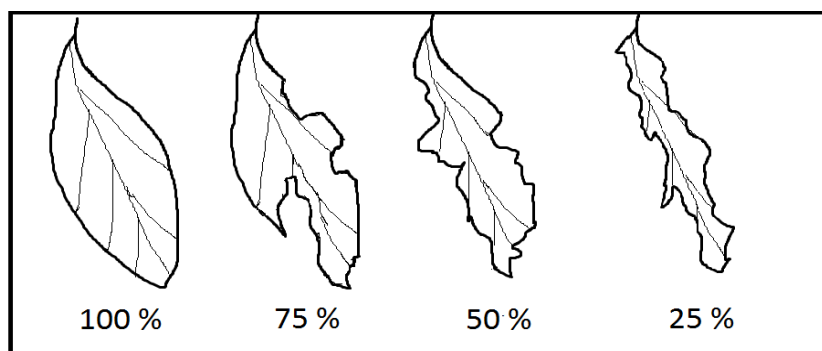
- large labelled plastic bag for transport to the drying ovens.
5. Dry the labelled paper bags in a drying oven at 70°C for 72 hours.
 6. Using laboratory scales, weigh the contents from each category in each trap separately.
 7. Record the results (in grams, to three decimal places).

Note:

- Leaf litter contents can be kept in a refrigerator for up to a week before being dried.
- If there is insect damage, leaf dry weight is likely to be low, biasing estimation of leaf productivity. Therefore, leaf loss needs to be quantified by sorting leaves from each trap into the closest matching category, and correcting for this loss as detailed in **Table 11** and **Figure 75**.

Table 11: Percentage loss categories of mangrove leaves

Category of leaf loss	Measured weight	Correction factor	Corrected weight
Full leaf		None (as measured)	
75% remaining		Multiply by 1.333	
50% remaining		Multiply by 2	
25% remaining		Multiply by 4	
Total for Trap =			

**Figure 75: Leaf percentage loss****17.7.2.1. Data interpretation**

Data is interpreted as mean dry weight of litter fall per square metre, per month ($\text{g}/\text{m}^2/\text{month}$). Ranges and standard deviations between traps should also be calculated.

Mangrove communities exhibit strong seasonal, annual and temporal variations in litter production, with peak fall occurring in summer in most locations, and varying with climatic conditions from year to year. It will also vary between regions due to different climate, rainfall, salinity and nutrient availability. For example, mangroves in the wet tropics are likely to produce more litter than those in more temperate climates. As different species also produce litter at varying rates, it is not usually appropriate to compare results between different regions and species.

Significant decline in leaf litter fall or reproductive effort in a particular mangrove community may indicate that it is under stress, so seek advice from relevant experts if such trends occur. If soil salinity is also being monitored, check data to see if unusual levels (high ones, in particular) have been recorded.

If comparing the stipules-to-leaves ratio in communities of *Rhizophora* spp., the ratio should normally be close to 1:1. Trends showing a higher ratio of leaves to stipules may indicate that the plants are shedding leaves, indicating possible stress.

For analysis of trends, data should be collected for at least three years, as some species of mangroves only produce propagules every two or three years.

17.7.3. Seedling regeneration

Seedlings should be monitored along the centre line of the plot at intervals of 10m in 1 x 1m plots.

The height and stem diameter of each seedling within the 1 x 1m plots are monitored every three months and used to calculate the growth rates of the seedlings. Stem diameters of seedlings will need to be measured at the same height above soil level on subsequent recordings.

Note: If seedlings are extremely dense the size of the seedling plots can be reduced to 0.5m².

1. Using the measuring stick, record the height of the seedling by measuring from the ground to the base of the uppermost apical shoot (**Figure 76**). Record the result on the datasheet.
2. Using callipers, measure the stem diameter of the seedling. This height will be determined by the height of the seedlings but 5cm above ground height would in most cases be an appropriate height.
3. For species that propagate using an elongated propagule rather than a seed (e.g. *Rhizophora* spp.) take the measurement at the base of the stem, just above the swelling of where the propagule meets the emerging stem (**Figure 76**).
4. Record density (number of stems per m²).
5. Count the number of leaves on each seedling. If there are more than 25, record the result as more than 25 leaves. Leaf counts provide an indicator of seedling progress in the early stages of development.
6. Collect a sample of the substrate at the middle of the transect and classify it based on McDonald and Isbell (2009).
7. It is also advisable to record the salinity and pH of the sediment within the transect.
8. Draw a mud map of the gap showing its dimensions, an arrow representing north, the position of the transect, and the surrounding forest type.

When the site is re-surveyed all seedlings, including any new seedlings, should have their height, stem diameter and leaf number recorded. Seedlings will only be needed to be measured up to a height of 1m.

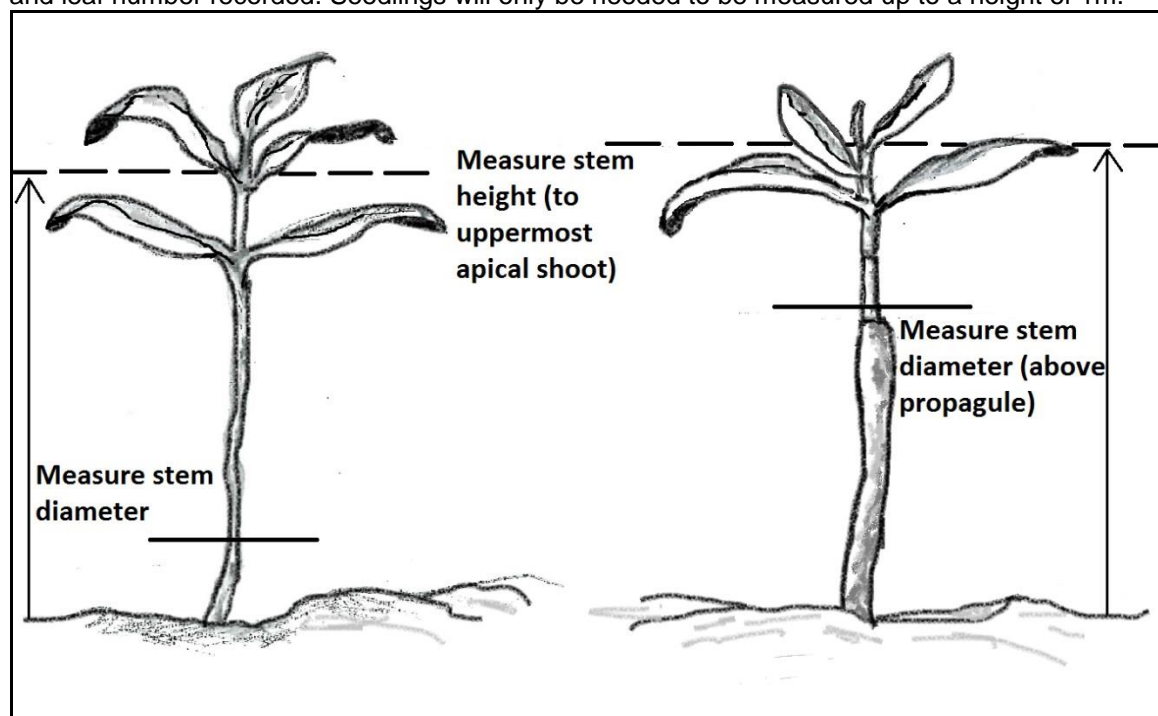


Figure 76: Where to take stem diameter and height measurement

17.7.4. Canopy cover and leaf area index

Light readings are taken in the sun using a light meter, outside the canopy of the mangrove community. A series of readings are then taken under the canopy, followed by a further series, again in the sun. Leaf area index (LAI) is determined by calculating the ratio of light under the canopy, to the light in the adjacent open space. The 50m x 10m quadrat is used.

Best timing is midday ± 2 hours to ensure that the sun is as close to overhead as possible. Valid measurements can only be made on sunny days. LAI measurement is not suitable for fringing mangrove environments, as light penetration from the edges will bias the result (e.g. on narrow river fringes). For more extensive mangrove communities ensure there are at least 20m between the quadrat and forest edge to avoid light penetration.

17.7.4.1. Light Meter Readings

Either a lux or photosynthetic active radiation (PAR) meter is suitable; the choice of meter depends on the accuracy required. Lux and PAR readings are not easily comparable, so choice of meter is important. Lux meters are cheap and robust, measure total light intensity, can be used to detect and measure short-term changes to mangrove canopy cover but have some limitations. In contrast, PAR meters are expensive and fragile. However, PAR meters measure photosynthetic active radiation (light absorbed by plants during photosynthesis) and are therefore highly sensitive and able to detect much smaller foliage pattern changes than the lux meter can.

To use the meter:

1. hold the light meter in one hand and the sensor in the other, ensuring that the white surface of the sensor is facing upwards
2. turn on the light meter and select a range that is appropriate for the current light conditions
3. to take a reading: blink your eyes and record the first reading that you see when you open them
4. record the results, including the range setting of the light meter (e.g. 1x, 10x and 100x)
5. wipe down the meter with a moist cloth after each use, treating the sensor with extreme care.

Note: Always read instruction manuals for light meters, as some require the application of a correction factor.

Take the light meter readings:

1. Outside the canopy:
 - 1.1. turn on the light meter and set the range to 100x
 - 1.2. take five readings outside the canopy (multiplying each by 100 to adjust for range)
 - 1.3. record the results.
2. Within the quadrat:
 - 2.1. set the light meter to 1x or 10x
 - 2.2. walk along the boundaries of the quadrat taking a light reading every metre for 100 m
 - 2.3. record the results (adjusting for the range).
3. Repeat step 1 above.

Note: When a light meter is used under a forest canopy, readings will occasionally go off-scale. If this happens, switch to a higher range setting and record the measurement on the new scale. Return to the original scale and continue to take readings. It is important to complete each set of readings within 30 minutes.

17.7.4.2. Measurement of the zenith angle of the sun

An instrument called a clinometer is used to measure the zenith angle of the sun, which is its angle from the vertical (**Figure 77**). The closer it is to midday, the smaller this angle will be. If a clinometer is not available, insert a 1-2m pole into a flat area of sunlit ground, ensuring it is vertical. Measure the height of the stick and the length of its shadow.

$$\text{Zenith angle} = \frac{\text{arc tan (length of shadow)}}{\text{height of stick}}$$

Alternatively, the zenith angle for the site can be calculated from a nautical almanac, a suitable computer program or phone/tablet application, using the latitude, longitude (or GPS reading) and time of day.

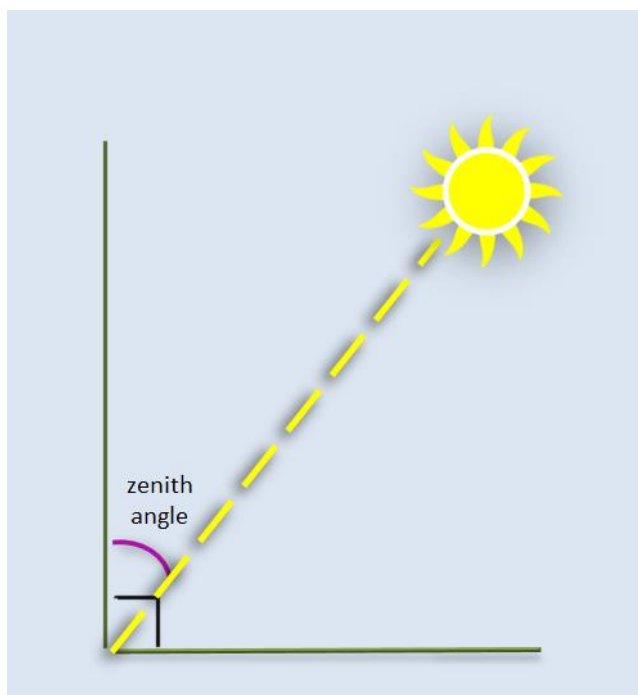


Figure 77: Zenith angle of the sun

17.7.4.3. Data interpretation

Calculate canopy cover and LAI of a plot using the following formulas:

$$\text{Canopy cover} = \frac{1 - \text{Average of canopy readings} \times 100}{\text{Average of open space readings}}$$

$$\text{LAI} = \frac{\text{Ln} \left(\frac{I_b}{I_0} \right)}{-k \times \cos \left(\frac{\infty \pi}{180} \right)}$$

Where:

Ln = Natural log of number

I_b = Mean value of light below the canopy

I_0 = Mean value of light above the canopy

k = Extinction coefficient that accounts for the angle and orientation of the foliage (a k value of 0.55 has been chosen as appropriate for mangrove stands).

∞ = Zenith angle of the sun

π = 3.14 (approximately)

Note: The k value quoted can be used in calculations for closed canopy forests of *Rhizophora*, *Bruguiera* and *Ceriops* spp. Due to the different structural characteristics of their canopies, it is **not suitable** for use in closed canopy forests of *Avicennia* spp., or in open forests. However, as no k value has, as yet, been calculated for *Avicennia* stands, the nominated value can be used to calculate LAI, but the data can be compared only with that from other *Avicennia* stands.

Forest LAI and canopy cover are the mean results from each plot. Data can be displayed on histograms as the LAI score, or as canopy cover per plot or forest over time. Median, range and standard deviations of readings are also calculated.

It is important to distinguish between natural and human-induced changes when interpreting data. As leaf area in canopies will naturally vary slightly from season to season, with a peak during the summer months, LAI can also vary naturally between sites and between different communities.

Large reductions in LAI are normally the result of disturbance or stress. If they are detected at a site, compare

results from a control or other site (containing the same species) to determine if this reduction is local or more widespread. It is also recommended to return to the site to observe the forest closely for evidence of damage (e.g. storm damage, insect attack or stress).

17.7.5. Crab burrow counts

The number of crab burrows in a survey area is estimated by counting burrows within 50 x 50cm quadrats. Monitoring should occur every three months.

This method is normally used in association with other methods, but if establishing a new site, ensure that it is in an area representative of the surrounding forest.

1. Establish three parallel 10m transects through the site, 5m apart. Mark the beginning and end of each with a peg to assist in locating the site again later.
2. Starting at 0m, place a quadrat to the left of the transect and count the number of crab burrows within it. Burrows on the edge of the quadrat should be counted only if the centre of the hole is within the quadrat.
3. If crab holes are very numerous, use a 25 x 25cm area of the quadrat and multiply the results by four.
4. Replace the quadrat and count crab holes every 2m along the length of the transect.

17.7.5.1. Data interpretation

Data is interpreted as crab holes per square metre (holes/m²).

Results may be highly variable between sites, so establish a baseline burrow density for each site. Long-term trends showing a significant decline in burrow numbers may indicate declining crab numbers and/or that the site is experiencing stress. Since crabs can have multiple burrow entrances and some species have been known to share burrows, the relationship is not linear.

Since crab hole abundance does not equate to absolute crab populations, significant changes in burrow counts would need to be recorded to indicate changes in population. Crab holes can be covered by sediment plugs at low tide.

17.7.6. Mangrove forest structure

This method is used to:

- provide baseline data on the diversity and structure of a mangrove community at a particular site,
- monitor long-term changes and provide a quantitative measure of species composition, stem density, and basal area of trees.

This information can be useful for interpreting other parameters, such as leaf trapping ability and LAI. Changes to basal area, stem density and canopy cover can be indicators of ecosystem health. It is a time-consuming method and should only be used to study long-term changes to mangrove forests.

1. Establish a transect running at right angles from the sea to the land, with 10 x 10m quadrats in each forest zone along the transect.
2. Use the compass to establish the bearing to follow.
3. Identify the major forest types or zones along the transect
4. For each forest type, find an area to the left of the transect that is representative (in terms of floristics and structure) of that mangrove community.
5. Within each quadrat, record the canopy cover, species type, tree height, sapling/seedling number and stem diameter.

Note:

- If two quadrats are to be established, ensure that they are at least 20m apart.
- If monitoring a homogenous forest type or a narrow mangrove fringe along a creek, transects can be established parallel to the shoreline. Quadrats can be placed where the forest is representative of the mangrove community, or at regular intervals.
- If there are a large number of trees or shrubs in the plot and the canopy within the plot is even, only record half the plot (but note which side of the plot it is). For plots with very large numbers of trees/shrubs (e.g. *Aegiceras* communities) it may be necessary to reduce the plot size.

17.7.6.1. Estimation of canopy cover

1. Walk along the centre line and longest edges of the plot and record where the canopy starts and finishes along

these lines. This includes recording breaks in the canopy along these lines.

2. Sum the distance that canopy covers along these lines.
3. Divide the figure by 3 and multiply by 2 to give a percentage cover for a 50 x 10m plot.
4. If the length of the plot is more or less than 50m you will need to adjust the figures accordingly.
5. If the canopy consists of more than one species of mangrove, estimate the percentage that each species contributes to the total canopy cover.

Note: Dominance is not the same as canopy cover; the total of all species must equal 100 per cent. For example, if there is a 70 per cent canopy cover and only one species, canopy dominance by that species is 100 per cent.

17.7.6.2. Measurement of stem diameter

1. Measure the stem diameter of each tree or shrub at breast height (1.3m above the ground) (**Figure 78**). Measure only those trees or shrubs with a height of 2m or more.
2. Record result as diameter at breast height (DBH) (**Figure 78**). A regular tape measure measures circumference only. Record this as circumference at breast height, and calculate DBH by dividing this result by π (approximately 3.14).
3. If carrying out long-term monitoring, hammer a galvanised nail (half of its length) into stems 10cm below where measurements have been taken, to provide a reference point for future measurements. Note this on the datasheet.

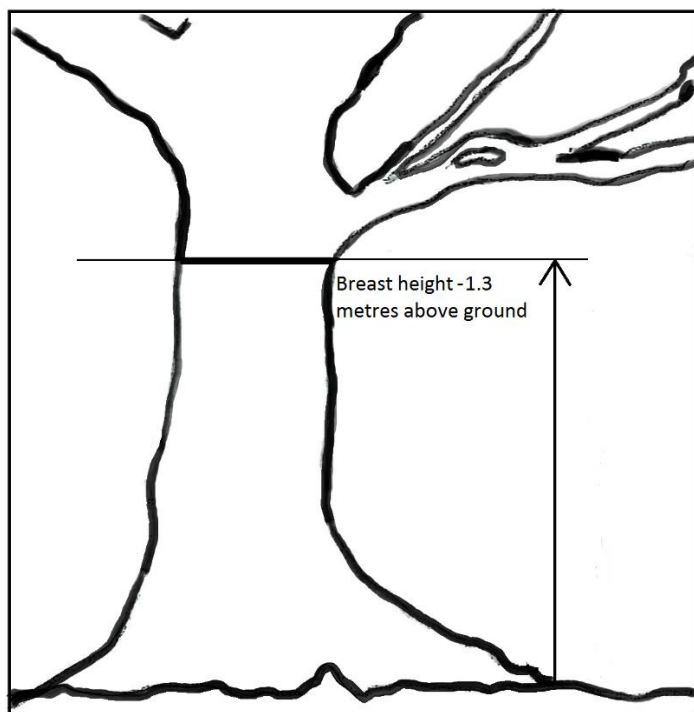


Figure 78: Measurements recorded at breast height

17.7.6.3. How to measure irregularly shaped trees

Irregularly shaped trees are very common in mangrove forests. If an irregularity occurs at breast height (**Figure 79**), use the following procedures to measure diameter:

- For multiple stems that fork below breast height; where stem diameter is 2.5cm or greater, measure the diameter of each stem at breast height, and record all results in the same box on the datasheet. Do not count each stem as a separate tree.
- For multiple stems that fork at breast height; take the measurement slightly below the swelling caused by the fork. For buttress roots, take the measurement 30cm above the uppermost prop root or buttress.
- For trunk swellings, take the measurement slightly above or below the swelling.

Some smaller mangrove forests may be naturally stunted or dwarf-like. In such situations these criteria are not suitable for determining growth status.

17.7.6.4. Saplings and seedlings counts

1. Count the number and record species type of seedlings and saplings within the quadrat.
 - 1.1. If plants are dense, use a smaller quadrat (size will depend on numbers, but 1 x 1m is a starting point), ensuring that the area sampled is representative of the larger quadrat.
 - 1.2. Estimate the number of seedlings/saplings within the 50 x 10m quadrat, based on the results of the smaller quadrat sampling.

17.7.6.5. Height estimation

To measure the height of each tree:

1. Stand the height pole up directly below the highest point of the tree (**Figure 80**).
2. Measure the height of the tree to the nearest 10cm, based on the known length of the pole.
3. Record the result.

Note: As this can be very difficult if the forest canopy is higher than 10m the use of a clinometer is recommended in such situations.

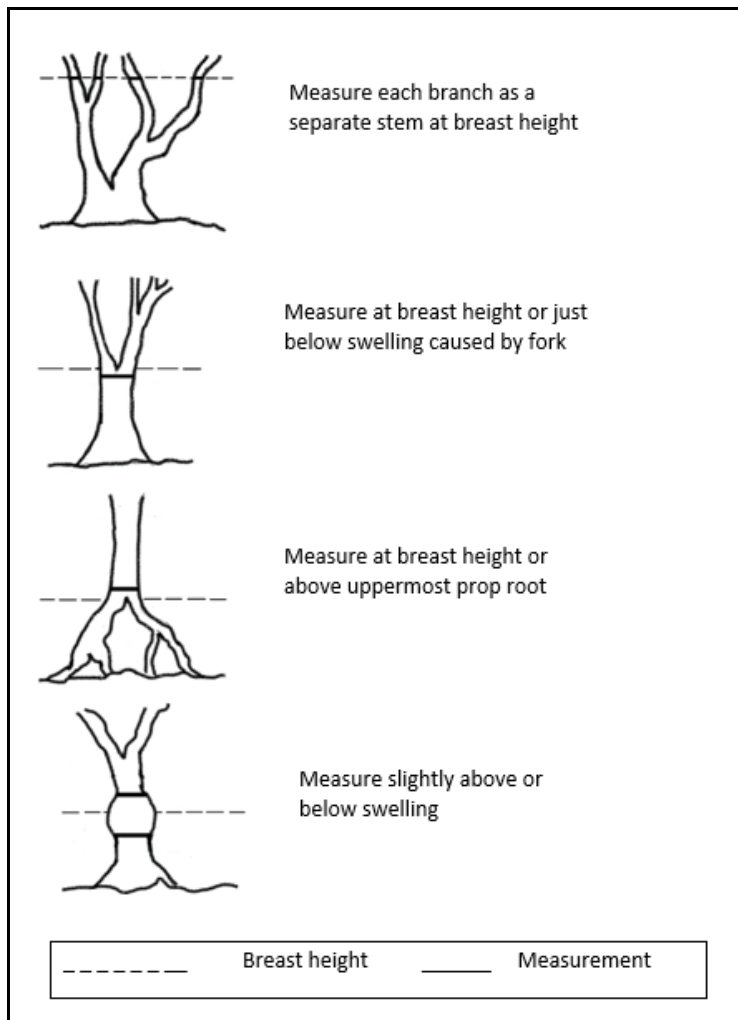


Figure 79: Measuring the stem diameter of irregularly shaped tree

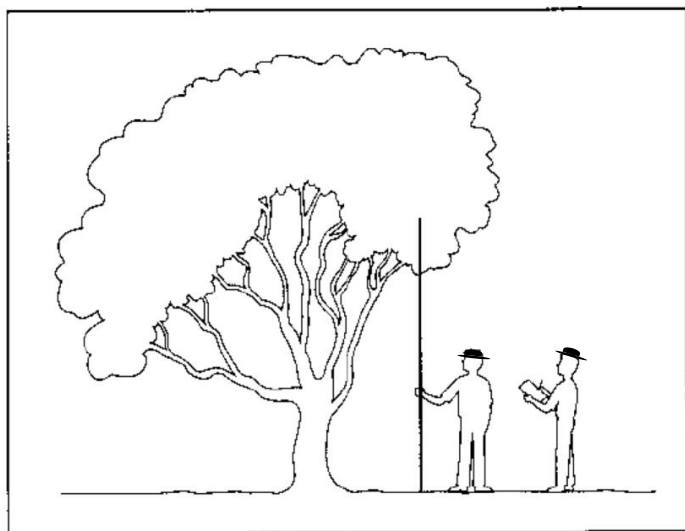


Figure 80: Using a height pole

17.7.6.6. Tag and record position of trees

Since branches can die during long-term monitoring of a plot, attach alloy or stainless steel tags to the main stem. Use nylon cable or stainless steel wire, ensuring that there is enough slack to allow for growth of the trees. The position of trees should also be recorded using a GPS.

17.7.6.7. Soils

Collect a sample of the substrate from the quadrat and rub it between your fingers. Record the sediment type based on its feel using the classification of McDonald and Isbell (2009). Other information such as pH and salinity can also be recorded.

17.7.6.8. Re-survey

As it is likely that a long time may have elapsed before repeat measurements are made, the original corner marker may have disappeared, but plot boundaries can be located using the tree tags. If new trees have become established, they should be assigned a new number.

17.7.6.9. Data interpretation

Formal measurements provide quantitative data on the structure or level of ecological development of a mangrove community. Data is expressed as:

- stems (living and dead) per hectare
- basal area (square metres per hectare)
- tree height.

Stems per hectare (stems/ha) is a measure of the density of living mangrove trees. It is calculated using the formula:

$$\text{Stems per hectare (stems/ha)} = \frac{\text{Number of living stems in plot} \times 10\,000}{\text{Area of the plot (m}^2\text{)}}$$

Stems per hectare should be calculated for each plot, together with the average for all the plots. The number of dead stems per hectare can also be calculated using the above formula, together with the overall ratio of dead to live stems (total dead stems versus total live stems).

Basal area (BA) of a plant refers to the cross-sectional area of its stem at 1.3m (breast height). The BA of a stand (stand BA) is the sum of all stem BAs in the quadrat, and is expressed as square metres per hectare (m²/ha). BA is a measure of the size, biomass or level of ecological development of a mangrove community. Normally, the higher the BA, the greater the biomass and level of development of a mangrove community.

Basal area for an individual plant is calculated using the following formula:

$$BA \text{ (cm}^2\text{)} = \pi r^2$$

Where:

$$r = \text{radius of the stem (cm)} = \frac{\text{DBH (cm)}}{2}$$

$\pi = 3.14$ (approximately)

If the plant has multiple stems, the basal area for the plant will be equal to the sum of the basal areas of the individual stems.

To calculate stand BA, use the following formula:

$$\text{Standard BA (m}^2\text{/ha)} = \frac{\Sigma \text{ BA for the plot (cm}^2\text{)}}{\text{Area of the plot (m}^2\text{)}}$$

Where:

Σ BA = sum of individual BAs

Increases in BA over time indicate that the community is still growing and developing. Increases in average canopy height will also help to confirm this. A significant decrease in BA may indicate that disturbance has occurred within the mangrove community. Average or median tree height can also be calculated to provide an indicator of canopy height and how the canopy is changing over time. Tree height measurements can also be used to track the progress of individual trees over time.

17.8. References and additional reading

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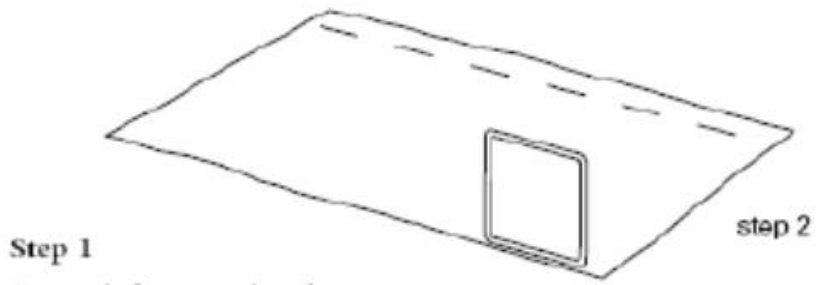
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Appendix 1

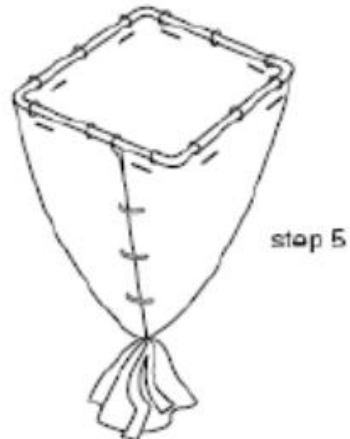
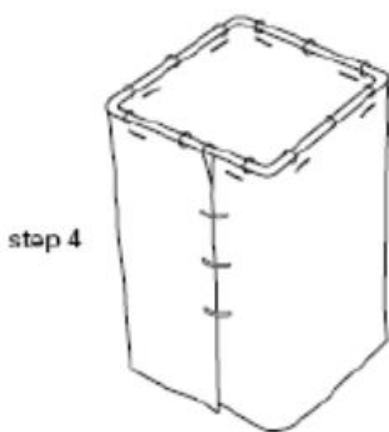
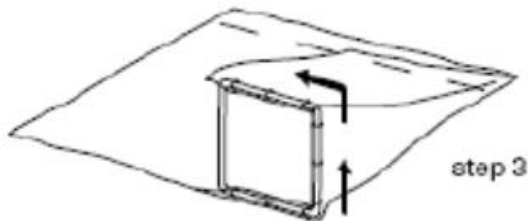
Table 1: Equipment checklist

General equipment for sampling in mangroves	✓	Mangrove litter trapping equipment	✓
Recent aerial photographs Waders 50m tape measure and shorter tape measure Mangrove species identification books Datasheets Waterproof Marker Pens GPS Camera		Leaf litter traps Nylon cord Marker pens Labelled paper bags Large labelled plastic bag Drying oven Laboratory scales	
Seedling regeneration equipment		Canopy cover equipment	
Compass Stakes 1 x1m square Shorter tape measure Flagging tape Measuring stick Plastic callipers Soil salinity and pH measuring equipment (recommended)		Light meter Moist cloth Clinometer or a 1-2m pole or GPS	
Mangrove forest structure equipment		Crab burrow counts equipment	
Compass 4 x PVC poles Light meter or forest densitometer Hammer Galvanised nail Height pole or clinometer Alloy or stainless steel tags GPS Callipers, tree callipers or diameter tape depending on trunk size Soil salinity and pH measuring equipment (recommended)		50 x 50cm quadrats. Marker peg	

Appendix 2 Construction of leaf litter traps



Cut each frame so that the internal trap dimensions will be 50 cm x 50 cm.



Background to sampling zooplankton

Version: February 2018

18. Background to sampling zooplankton

18.1. Purpose and scope

This document provides background information on sampling zooplankton.

18.2. Associated documents

Biological assessment: Sampling zooplankton with a Schindler-Patalas trap

Physical and chemical assessment: Manual collection of surface water samples (including field filtration)

18.3. Introduction

Zooplankton comprise a complex group of predominantly microscopic animals such as radiolarians, foraminifera, tintinnids, thecate amoeba, amphipods, krill, copepods, cladocerans but also include the immature stages of larger animals such as animals such as sea urchins, many crustaceans, marine worms, marine snails and most fish.

These organisms can be found free floating, (drifting) in the water column of non-marine (freshwater), brackish and fully marine environments. They are an essential component of all aquatic food webs because they provide the link between the primary producers (bacterioplankton, phytoplankton) and larger animals such as fish.

Zooplankton species occupy particular niches within their aquatic habitats (ponds and lagoons, rivers and streams, lakes and estuaries, open ocean) and are influenced by a number of environmental factors. These include light, temperature, turbulence, and salinity. Zooplankton are also directly and indirectly affected by pH, dissolved and particulate metals (whether present as essential trace elements or as toxic contaminants) and the availability of food. Nutrients such as nitrogen and phosphorus can also have an indirect impact on zooplankton growth, because growth of phytoplankton (which forms the primary food source for zooplankton) is affected by the availability of these nutrients in the water, which in turn impacts on zooplankton survival (bottom-up control). Zooplankton populations are also controlled through predation (top-down control). Both phytoplankton and zooplankton abundance and species diversity are therefore highly responsive to environmental conditions ranging from nutrient availability, temperature, light, pollution, food quality and the degree of predation by other animals.

Changes in zooplankton communities over time may indicate subtle environmental changes to their habitats. Therefore, indices of zooplankton abundance and species diversity can be used to determine the health of an ecosystem. Many local councils and water managers collect phytoplankton and zooplankton samples as indicators of water quality, often in response to the increasing incidence of algal blooms in rivers and estuaries.

18.4. Sampling zooplankton

The most common method for sampling zooplankton is by dragging a fine mesh net, either horizontally or vertically through the water. This method can filter a large volume of water, but the results will only be qualitative or at best, semi-quantitative, despite the availability of standard methods that specify sampling procedures such as rope length to be used and time of net drag. For quantitative results in surface waters, a Schindler-Patalas zooplankton trap can be used. Because the trap holds a known quantity of water, the number of zooplankton per litre can be calculated. It is almost entirely constructed of transparent material, which helps prevent avoidance reactions from some planktonic organisms. The trap is also light-weight and easily operated by a single operator, however can only be used to collect samples in the top 60–70 cm of a water body. If quantitative samples are to be collected from a discrete depth/s, a Van Dorn water sampler or a Niskin bottle can be used as for water sampling (see Manual collection of surface water samples (including field filtration) document). Water in the water sampler is filtered from the water sampler through a plankton net/sieve with appropriate mesh size (based on the size of the target organisms).

18.5. References and additional reading

Baranyi, C, Hein, T, Holarek, C, Keckeis, S and Schiemer, F 2002, 'Zooplankton biomass and community structure in a Danube River floodplain system: effects of hydrology', *Freshwater Biology* 47, 1-10.

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- Shiel, RJ, Walker, KF, Williams, WD 1982, 'Plankton of the Lower River Murray, South Australia', *Australian Journal of Marine and Freshwater Research* 33, 301-27.

Sampling zooplankton with a Schindler-Patalas trap

Version: February 2018

19. Sampling zooplankton with a Schindler-Patalas trap

19.1. Purpose and scope

This document describes the sampling of zooplankton communities using a Schindler-Patalas zooplankton trap.

19.2. Associated documents

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

Biological Assessment:

- *Background to sampling zooplankton*

19.3. Health and safety

Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

19.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

19.5. Skills, training and experience

Skills, training and/or experience to undertake this method should include at least one member of the sampling party with previous training and experience in the use of this method.

19.6. Equipment

See Appendix 1 for example equipment checklist.

19.7. Procedure

19.7.1. Preparation for sampling.

Choose a suitable sized Schindler-Patalas zooplankton trap: The size of the trap will depend on the study aims.

Choose an appropriate mesh size: The mesh size used will be based on the objectives of the study (i.e. the size of the target organisms).

19.7.2. Shallow water

- 1 Prior to entering the water, the height of the trap should be measured against the operator's body. For the trap to operate correctly, the water depth must be greater than the height of the trap.
- 2 Wade into the water (with the trap out of the water) to a depth that is adequate for sampling. For example, if the trap height is equal to the user's hip height, the trap can be operated when the user is standing in water up to their hip.
- 3 Lower the trap into the water at an angle perpendicular to the substrate. As the trap enters the water, the hinged doors, top and bottom, open upwards as water (and zooplankton) flows through. Lower the trap until it is below the water level.



- 4 Once the trap is full of water, raise it above water level. Water will flow through the netting at the bottom of the trap, and zooplankton will flow into the small sampling container attached at the bottom of the trap.



- 5 Tilt trap slightly so that the entire sample flows out of the trap and through the net to the collecting container.



- 6 Wash the sides of the netting down with water from the river.



- 7 Open the bottom of the sampling container and pour the contents into an appropriate vial. The contents should be approximately 20mL.



- 8 Label the vial and add a preserving agent (e.g. 100% methylated spirits or ethanol) if required. The amount used should be enough to have a final concentration of preservative in the sample of approximately 80%.
- 9 Record the site name, sample name, date and time and samplers name. Record the volume of the sampler.
- 10 Store sample in an upright position in a safe and secure storage place until it is submitted to experts for analysis.

19.7.3. Deep water

In deep water, the Schindler-Patalas trap can be used to sample zooplankton from a boat. The procedure is the same as in shallow water, except the person places the trap into the water from a boat.

19.7.4. Replicate samples

Zooplankton community distribution tends to be patchy, varying horizontally and vertically. Spatial heterogeneity is a function of density, taxonomic species, body size, developmental stage, season, and thermal stratification (Pinel-Alloul et al. 1988). The acceptable number of replicates generally required for zooplankton sampling with a trap in rivers is between two and four (Shiel et al. 1982, Evans and Sell 1983, Downing et al. 1987, Baranyi et al. 2002, King 2004).

19.7.5. Limitations

- The Schindler-Patalas trap can only be used in water deeper than the trap length (approximately 60-70cm).
- Sampling using the Schindler-Patalas trap is limited to the dimensions of the trap and the number of samples taken. Therefore, depending on the spatial distribution of zooplankton at a given time, a large number of samples may need to be taken to detect zooplankton assemblages, as opposed to pulling a net through the water for a given distance/time, so that a large volume of water can be sampled in a short time.
- The Schindler-Patalas trap is designed for collecting zooplankton samples from waters surface (to a depth of 60–70cm).

19.8. References and additional reading

Baranyi, C, Hein, T, Holarek, C, Keckeis, S and Schiemer, F 2002, 'Zooplankton biomass and community structure in a Danube River floodplain system: effects of hydrology', *Freshwater Biology* 47, 1-10.

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Seda, J and Devetter, M 2000, 'Zooplankton community structure along a trophic gradient in a canyon-shaped dam reservoir', *Journal of Plankton Research* 22, 1829-1840.

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Shiel, RJ, Walker, KF, Williams, WD 1982, 'Plankton of the Lower River Murray, South Australia', Australian Journal of Marine and Freshwater Research 33, 301-27.

Appendix 1**Table 1 Equipment checklist**

Equipment	✓
Schindler-Patalas zooplankton trap:	
Mesh net for zooplankton trap:	
Plastic vials/jars	
Waterproof pen/pencil for labelling	
Bucket (Optional)	
Methylated spirits 100%, or Ethanol	
Pipettes (to add preservative)	

Background information on freshwater and marine microalgae and harmful algal blooms (HABs)

Version: February 2018

20. Background information on freshwater and marine microalgae and harmful algal blooms (HABs)

20.1. Purpose and scope

This document provides background information on freshwater and marine microalgae and harmful algal blooms (HABs).

20.2. Associated documents

Biological assessment:

- *Sampling freshwater and marine microalgae and harmful algal blooms (HABs)*
- *Physical and chemical assessment - Chlorophyll a sample collection methods*

20.3. Introduction

Freshwater and marine microalgae, also known as phytoplankton, are an important group of microscopic plants for monitoring water quality because of their sensitivity to environmental changes. Microalgae are therefore good indicators of aquatic health because they reflect changes in water quality and/or environmental degradation.

In general, the greater the diversity of phytoplankton in a water sample, the better the health of the water body. If the number and diversity of phytoplankton genera and species is seen to decline during a routine monitoring program, this could be either the result of natural seasonal fluctuations or indicate that the water quality is declining in that water body.

There are many types of phytoplankton (**Figure 81** and **Figure 82**) which can be divided into four main groups—the blue-green algae (cyanobacteria, which are not strictly algae), green algae, diatoms and flagellates (the latter including dinoflagellates and raphidophytes).



Figure 81: Green microalgae (a) Chroococcus turgidus (b) Micrasterias. Photo credits: Glenn McGregor, DES

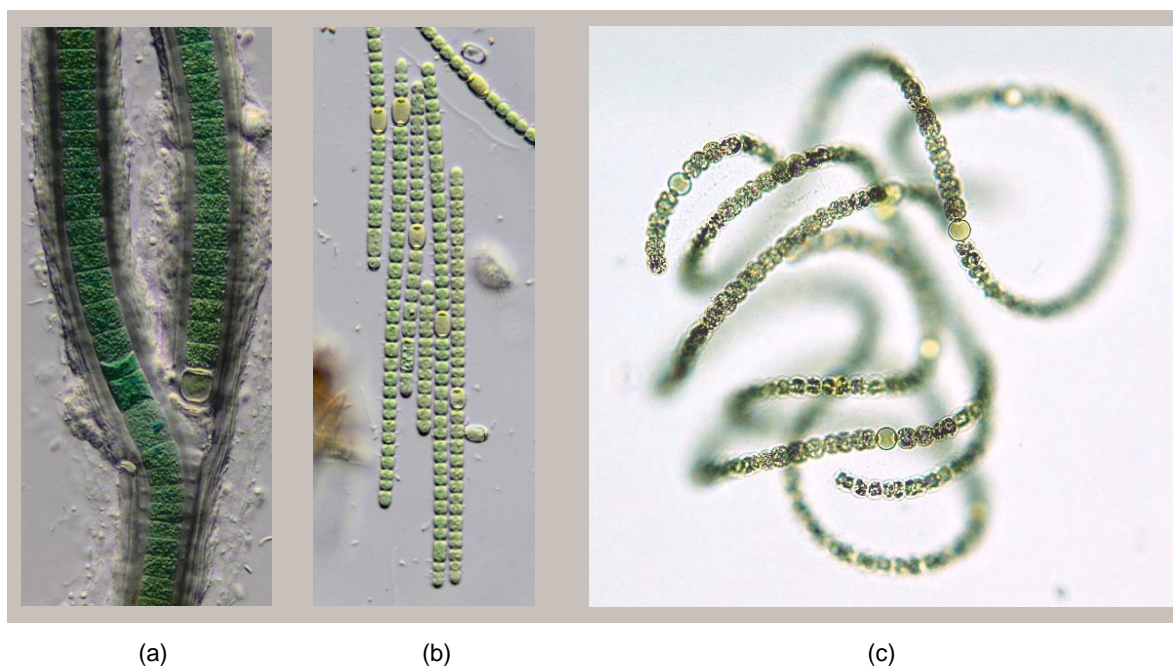


Figure 82: Blue-green algae (a) Tolypothrix sp. (b) Anabaena sp. (c) Dolichospermum circinale. Photo credits: Glenn McGregor, DES

Algae are important components of any aquatic environment because they are the primary producers at the base of all aquatic food chains. In addition to this, being photosynthetic “plants” they utilise dissolved CO₂ in the water and release oxygen, which meets the respiration needs of aquatic animals.

A number of environmental factors (including nutrients, light, water temperature, pH, salinity and turbidity) influence the growth of phytoplankton in aquatic environments. In some instances when excessive nutrients are available, phytoplankton can proliferate and form large algal blooms that are usually visible on the surface of a waterbody (**Figure 83**). Phytoplankton biomass in a water sample can be estimated by the photosynthetic pigment chlorophyll a which is found in all phytoplankton cells.

Algal blooms may occur under natural conditions and many are beneficial to aquatic environments by providing food sources for other organisms. However, increased frequency and intensity of algal blooms may be attributed to increased nutrient levels entering the water from either diffuse or point-source discharges (e.g. sewage treatment plants, agricultural irrigation, industrial effluents and domestic activities). Although phytoplankton/microalgae produce oxygen during photosynthesis and take up nutrients, large algal blooms can result in poor water quality. This is because the cells of the algal bloom eventually die and the subsequent microbial decomposition has the potential to deplete oxygen dissolved in the water. If oxygen concentrations drop low enough, it can lead to the death of other aquatic organisms, including fish and macroinvertebrates. Any bloom of sufficient biomass relative to the size of the waterbody can produce this harmful effect. This is an example of a non-specific harmful algal bloom (as it can be caused by species that are otherwise harmless).

20.4. Harmful algal blooms- freshwater blue-green algae (cyanobacteria)

Harmful Algal Blooms (HABs) are common, sometimes seasonal phenomena that occur throughout Queensland in fresh, estuarine and coastal marine waters. Their occurrence has the potential to seriously degrade water quality. When the total cell counts reach excessively high numbers they become evident as discoloured water, usually green or blue-green in colour, frequently with a strong musty odour and visible thick surface scums (**Figure 84**).

The organisms largely involved in these harmful outbreaks include cyanobacteria (such as *Microcystis*, *Dolichospermum*, *Nodularia*, *Cylindrospermopsis*, and *Chrysochloris*). Harmful algal blooms may produce toxins that pose a direct threat to human and animal health, or otherwise adversely affect water quality through the production of taste and odour compounds that reduce the suitability of water for direct consumption or recreational activities. Consequently, harmful algal blooms may have economic impacts from increased costs of water treatment or the need to use an alternative water supply, as well as social impacts through disruptions to the recreational use of the waterway. Toxin-producing HABs may also have deleterious effects on aquatic ecosystems.



Figure 83: Photographs of fresh water algal blooms in lakes and a small farm dam. Photo credits: Philippa Uwins, DES





Figure 84: Fresh water cyanobacteria blooms. Photo credits: Glenn McGregor, DSITI

Recent research has also linked a neurotoxin known as BMAA (β -methylamino-L-alanine) that can be produced by a range of cyanobacteria (as well as some marine diatoms and dinoflagellates) as a possible contributing factor to neurodegenerative disease (Cox et al. 2016).

The Queensland Harmful Algal Bloom Response Plan (DNRM 2014) outlines the contingency plan for responding to HABs within the capacity of local response agencies such as the Department of Natural Resources and Mines (DNRM), Department of Environment and Science (DES), Department of Health (DoH), Department of Agriculture and Fisheries (DAF), Department of National Parks, Sport and Racing (NPSR), local governments and water storage operators.

As described above, blue-green algal blooms form clearly visible surface scums which can be highly toxic. In view of this, extreme care should be taken when sampling water (e.g. the use of implements to collect samples, wearing protective long sleeved safety gloves etc.) in order to avoid any body contact (See detailed procedures in Sampling freshwater and marine microalgae and harmful algal blooms document for harmful algal bloom sample collection and handling).

Reporting blue-green algal blooms is particularly important because of the wide range of users that can be affected. Rapid reporting to the responsible water authority will allow the authorities to alert their aquatic plant experts to verify the identification and perform a cell count of the numbers of organisms, so that control procedures can be implemented and appropriate signage put up to alert the public.

20.5. Marine harmful microalgae

Around the world a range of marine microalgae can cause harmful algal blooms and these are sometimes known as 'red tides', although most blooms don't have a red colour. The most commonly occurring harmful marine algae in coastal Queensland is *Lyngbya majuscula* (a large filamentous cyanobacterium or blue-green algae) (**Figure 85**). Toxicological tests from Moreton Bay have identified toxins that may occur separately or in combination in *Lyngbya majuscula* populations. The toxins identified from Queensland populations are Lyngbyatoxin A and Debromoaplysiatoxin.

Another potentially harmful group of marine cyanobacteria that commonly occur in Queensland waters are species of *Trichodesmium*, which have been linked to skin irritations in humans. Large, floating drifts of *Trichodesmium* are commonly seen on the surface of estuarine and coastal Queensland waters during periods of calm weather (**Figure 86**).

The other main group of microalgae responsible for marine HABs around the world are flagellates, especially dinoflagellates and raphidophytes. Certain species of these groups have been responsible for fish kills on a large scale. In addition, toxins produced by some species of dinoflagellate (and diatom) can accumulate in seafood (especially shellfish) to cause human poisoning. In Queensland, the most common form of seafood poisoning caused by marine microalgae is ciguatera, from consumption of normally edible species of fish contaminated with ciguatoxins. Ciguatoxins are produced by benthic dinoflagellates belonging to the genus *Gambierdiscus* (Holmes et al. 2014). *Gambierdiscus* are common epiphytes of seaweeds on coral reefs along the coast of Queensland (Gillespie et al. 1985) and the ciguatoxins they produce can accumulate through marine food chains into normally edible species of fish.



Figure 85: (a) Large blooms of *Lyngbya majuscula* smothering seagrass beds in Moreton bay. Photo credit: DES, Queensland Government Wetland Info. (b) *Lyngbya* bloom washed onto foreshores in Deception Bay and smothering mangrove roots (Photo credit: Moreton Bay Regional Council)



Figure 86: *Trichodesmium* bloom

20.6. Shellfish poisonings caused by HABs

Shellfish can accumulate and concentrate phytoplankton toxins from a range of marine dinoflagellate and diatom species. To date, such shellfish poisonings have been rare in Queensland. Different HAB species produce different toxins that cause different types of seafood poisonings such as Diarrhetic Shellfish Poisoning (DSP), Paralytic Shellfish Poisoning (PSP), Neurotoxic Shellfish Poisoning (NSP) and Amnesic Shellfish Poisoning (ASP). In some cases, shellfish can become poisonous when relatively low numbers of HAB species are present in the water.

20.7. Stock and wildlife affected by harmful microalgae

Stock animals are often affected by algal blooms through the consumption of freshwaters contaminated with cyanotoxins (toxins produced by blue-green algae). All animal species are potentially susceptible. Fatal cases are on record for cattle, sheep, horses, pigs, dogs and birds. Wildlife most commonly affected by harmful algae are fish (**Figure 87**) and water birds.

A range of marine flagellate species can also produce toxins that can kill fish and other wildlife. The flagellate species most often associated with fish kills are certain dinoflagellate and raphidophyte species.



(a)

(b)

Figure 87: (a, b) Algal bloom, Caboolture River with fish kill

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Sampling freshwater and marine microalgae and harmful algal blooms (HABs)

Version: February 2018

21. Sampling freshwater and marine microalgae and harmful algal blooms (HABs)

21.1. Purpose and scope

This document outlines standard procedures to collect and preserve freshwater and marine microalgae from aquatic environments. General methods are also described for safely collecting and handling water samples of known or suspected harmful blue-green algae (cyanobacteria) that may contain toxins.

This document does not provide information on the collection of algae samples for toxin analysis, nor the collection of tissue samples of livestock, wildlife, fish or shellfish that are suspected to have been in contact with, or have ingested harmful algal toxins. These methods are provided in Queensland Harmful Algal Blooms Operational Procedures (DNRM 2014). The Queensland Harmful Algal Blooms Operational Procedures also describes how to report suspected blue-green algal blooms to the appropriate authorities.

21.2. Associated documents

Biological assessment: Background information on freshwater and marine microalgae and harmful algal blooms (HABs)

Sampling design and preparation:

Permits and approvals

Record keeping including taking field photographs and videos

21.3. Health and safety

Some algal species can cause health issues. Before following the methods contained in this document, a detailed risk management process (identification, assessment, control and review of the hazards and risks) must be undertaken. All work carried out must comply with the Queensland Work Health and Safety legislative obligations.

21.4. Permits and approvals

Permits and approvals may be required to conduct activities involving animals, plants and/or in protected areas (for example National Park/Regional Park, State Forest or State Marine Park). See Permits and approvals document for more information on requirements.

21.5. Skills, training and experience

Staff using this method should have previous training or experience in:

- recognising algal blooms and associated surface scums
- how to collect, transport and deliver samples to a laboratory for analysis.

21.6. Equipment

See Appendix 1 for equipment check list and Appendix 2 for preparation of Lugol's solution.

21.7. Procedure

21.7.1. Preparation for sampling

Note: Samples should be collected in 500-1000mL plastic or glass sampling bottles.

21.7.1.1. Ecosystem surveys

When undertaking ecosystem surveys:

Be prepared to collect samples that represent a range of habitats

Prepare to collect water samples at a particular site at approximately the same time of day each time, preferably between 8.30am and 12.00pm. This is because the algae move up in the water towards the surface in the morning, and tend to sink to lower regions in the afternoon. By sampling at roughly the same time on each occasion the survey results can be directly compared over time.

21.7.1.2. Algal blooms

Algal blooms (including cyanobacterial blooms) can be extremely patchy in distribution, both spatially and temporally. Buoyant species tend to accumulate near the surface or along the shoreline at the downwind or downstream end of reservoirs or river reaches. In view of this, "depth integrated" open water sampling is the preferred option, because it provides a better representation of the "true" or average algal population in a water body.

When monitoring algal blooms, the selection of sampling sites will depend on a number of factors, including:

- the time of day
- prevailing winds
- proximity to tributary inflows i.e. where a stream flows into a larger body of water
- the proximity to potential nutrient input sites.

Collect samples at any time if an algal bloom has been reported.

Note: Some species of blue-green algae can cause skin irritation. If sampling from an area that has a high level of phytoplankton, wear gloves and appropriate protective clothing to minimise contact with the water. Where skin contact does occur, the skin should be washed immediately.

21.7.2. Collection of algae samples from a stream or small river

21.7.2.1. Collecting samples directly from a stream or river

- 1 Choose a site that is representative of the bulk water being assessed.
- 2 If sampling an algal bloom, take notes that describe the surface scum (colour, odour, presence of dead organisms etc.).
- 3 Label sample bottles with a water proof pen.
- 4 Enter the water, move towards midstream and face upstream to collect the sample (**Figure 88**).
- 5 If the bottle needs rinsing (i.e. if reusing bottles from previous samplings) wash the sample bottle with ambient water three times, discarding the rinse water downstream.
- 6 To fill the sample bottle the following method should be used to avoid sampling surface scum.
- 7 Grip the bottle in one hand around the base and remove the lid with the other hand.
- 8 Invert the sample container fully and submerge to a depth of 0.2m below the surface.
- 9 Turn the mouth of the bottle upwards and towards the current.
- 10 When the bottle is full remove it from the water rapidly and replace the lid. Take care to keep fingers clear of the lid liner and neck of the bottle.
- 11 If sampling an algal bloom, surface scums can be targeted by moving the sample bottle through the surface.
- 12 If preservation with Lugol's solution is required to stain and preserve the sample, add a couple of drops of Lugol's solution into the sample to preserve it. The sample should turn the colour of weak tea.
- 13 Check that details on the sample container are correct.
- 14 Place the sample bottle(s) in a cool box (with ice or ice bricks) or portable refrigerator that is suitable for transport to the analysing laboratory. Record the site, sample name, date and time, sampler name and any general observations about the site.

Note: Accumulations of surface scums are useful for identification purposes, but once diluted they should not be used for counting purposes (quantitative analysis).



Figure 88: Collecting a grab sample

21.7.2.2.

Collection of samples using a pole-type sampler

1. Follow steps 1–3 in Section 21.7.2.1.
2. Extend the sampling pole to the required length and check that all surfaces have been cleaned.
3. Remove the lid of the sample container.
4. Place the sample container into the bracket of the sampling pole.
5. Invert the sample container and submerge to a depth of 0.3m below the water surface (**Figure 89**).
6. Rotate the sample container into the direction of flow and fill the sample bottle.
7. Turn the sample bottle upright, remove from the water and replace the bottle lid. Take care to keep fingers clear of the lid liner and neck of the bottle.
8. Follow steps 11-14 in Section 21.7.2.1.



Figure 89: Using an extendable pole type sampler**8.1.1. Collection of algae samples from large rivers, lakes or the ocean**

Samples in large rivers, estuaries, oceans and lakes can be collected using a hose-pipe sampler (**Figure 90**) or using a depth sampler, e.g. Van Dorn (**Figure 91**).

Note:

When sampling from a boat, rinse the bottle on the side of the boat opposite from where samples are collected in order to avoid disturbance of the surface algal community.

When a fuel-powered boat is used, collect samples from the bow to avoid contamination from the motor.

8.1.1.1. Hose-pipe sampler (for depth integrated samples)

A hose pipe sampler is a weighted hose-pipe that can be used to obtain a water column sample. It is usually used for collecting depth integrated samples when a representative sample of the water column is desired. A hose-pipe sampler can be constructed using a garden hose. The length of the hose-pipe should be chosen to reflect the appropriate depth to which the cells are likely to be mixed. A temperature probe can be used to determine the mixing status of the waterbody and depth of any thermocline present. Water samples can then be collected from the surface to the thermocline.

Note: It is possible to use a rigid pipe fitted with a one-way valve, instead of an integrated hose-pipe sampler. It can simplify the operation of withdrawing the pipe and sample from the water.

- If mixing status is unknown, a 5m long hose-pipe is recommended.
- At shallow sites (less than 3m water depth) a 2m long hose-pipe should be used.
- At deeper sites longer hose-pipes (up to 10m) may be useful.

The method for collecting samples using a hose-pipe sampler is shown in **Figure 90**. The procedure for collecting a sample using an integrated hose-pipe sampler is:

- 1 Determine the depth of any thermocline present using a temperature probe and collect samples from the surface to the thermocline.
- 2 If you do not have a temperature probe use a 5m long hose-pipe as a default.
- 3 Attach a cord to one end of the hose and the boat to prevent accidental loss of the hose-pipe.
- 4 Holding the hose-pipe at the top end, rapidly drop the weighted end of the hose-pipe into the water to a depth of approximately 5m.
- 5 Pull the bottom of the hose-pipe into the boat using the cord without inserting the rubber cork.
- 6 Rinse the hose-pipe.
- 7 Repeat steps 3-4, but this time insert the cork into the top end of the hose-pipe before pulling the hose-pipe into the boat.
- 8 Pull the bottom end of the hose-pipe to the surface using the cord, so that the tube is in a U-shape (**Figure 90**).
- 9 Lower the weighted end of the hose into a bucket and remove the cork. Ensure that the entire contents of the hose are emptied into the bucket.
- 10 Mix the contents of the bucket and then transfer part of the contents into a sample bottle. Discard the rest of the contents of the bucket.
- 11 Record the site, sample name, date and time, sampler name, depth and any general observations about the site.
- 12 Clean the hose-pipe and sampling bucket (rinse several times thoroughly with clean water).

When not in use, the hose-pipe sampler and bucket should be kept clean and stored in a dark shed or cupboard.

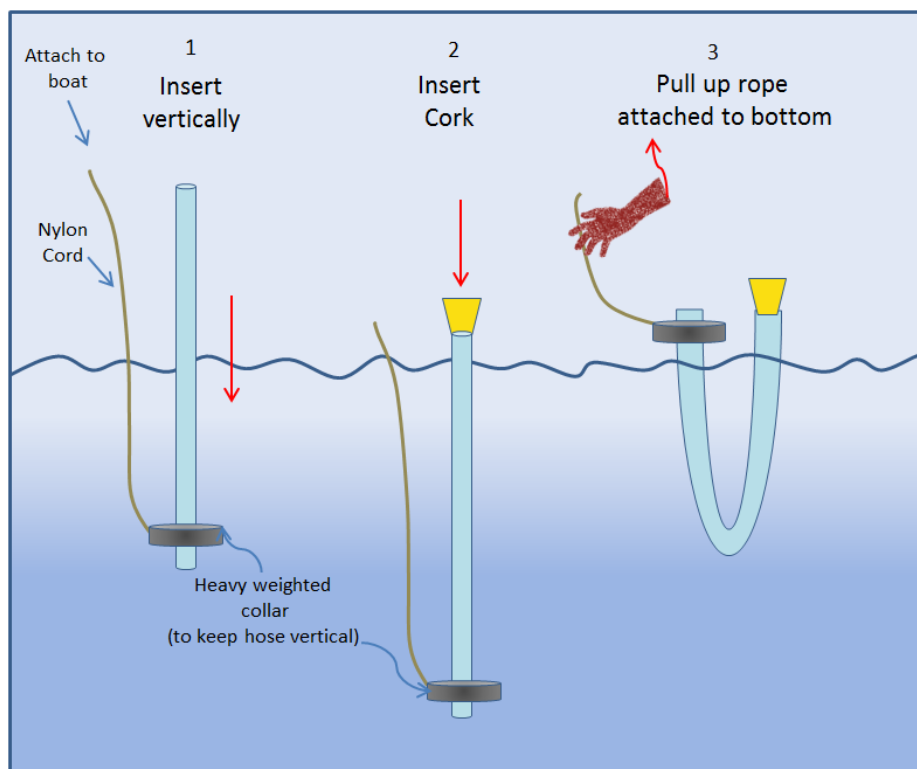


Figure 90: Procedure for use of the integrated hose-pipe sampler

8.1.1.2. Collecting samples using a depth sampler

Samples can be collected at discrete depths using a sampler such as a Van Dorn sampler (Figure 91) or Niskin bottles. Some Niskin bottles are fitted with a thermometer to record the temperature of the water at the sampling location.

- 1 The general procedure for collecting samples using a depth sampler is:
- 2 Cock the sampling bottle as per the manufacturer's instruction.
- 3 Lower the bottle into the water to the required depth.
- 4 Trigger depth sampler as per the manufacturer's instruction.
- 5 Remove the device from the water.
- 6 Gently transfer the volume required to labelled sample bottles.
- 7 Record the site, sample name, date and time, sampler name depth, and any general observations about the site.
- 8 Fix the samples using Lugol's solution (if required).
- 9 Rinse the sampling bottle thoroughly between each site.



Figure 91: Van Dorn sample bottle

8.1.1.3. Preservation and transport of microalgae samples

Samples should be examined as soon as possible after collection while the algae are still alive, as some identifying

features are more clearly seen in live algae. Consult with the analytical laboratory as to their preferred method for maintaining, preserving and transporting live samples. For example, unpreserved samples received by the laboratory greater than 48 hours after collection may not be analysed. Algal samples should be kept cool or cold on ice in a cool box or in a portable refrigerator and stored in the dark until samples can be examined. Samples should not be allowed to freeze.

Samples may be preserved using Lugol's solution if required. Lugol's solution can be added to the water samples drop by drop, using a disposable pipette until the sample is a weak tea colour (i.e. approx. 0.5mL Lugol's solution to 100mL of sample). If there is a high concentration of algal cells (such as in a bloom event), it may be necessary to add more Lugol's solution to the water sample. Lugol's solution is commercially available or can be prepared easily as described in Appendix 2.

Note: Algal samples may be considered 'dangerous goods'. Commercial carriers have shipping regulations—ensure the sample packaging and labelling meet the requirements. Contact the courier company for details prior to sampling.

8.1.2. Freshwater and marine benthic microalgae

Filamentous and other benthic algae can usually be found attached to substrates such as rocks, sand and **gravel** beds, woody debris, buoys and mooring fixtures where they form mats. These attached algae can be collected for taxonomic and qualitative analysis as mats or strands of filaments.

Note: Samples should be collected in a high density polyethylene screw top jar and filled with surrounding water and fixed with Lugol's solution (if required).

8.1.2.1. Collecting samples from shallow water

Small samples from shallow near shore sites can be collected by hand or scrapers/spatulas (sample size around the size of a ten cent coin). Larger samples can be collected using nets or rakes, ensuring that the full varieties of habitats present at a site are sampled. Duplicate samples across the area of interest should be collected.

If there is little variation in the type of material across the area of interest, a composite sample may be taken incorporating multiple subsamples.

8.1.2.2. Collecting samples from deep water

Benthic algae samples in deep water sites using a benthic sampler such as a Van Veen grab sampler (**Figure 92**) or an Eckman grab sampler.



Figure 92: Van Veen Grab Sampler collecting benthic mud in sample container

8.1.2.3. Preservation and transport of benthic algae samples

Samples from different habitats and substrates should be separately bottled or bagged.

- 1 Refrigerate or chill immediately after collection (to 4°C) but do not allow the sample to freeze. Samples will remain fresh for approximately two days.
- 2 When ready to send for analysis, place the sample jar into a zip lock bag and put it into a small plastic cool box with one or two freezer bricks.
- 3 Pad the spaces with absorbent material in case the sample leaks and tape up the package securely. Send the

samples as soon as possible.

Note: Algal samples may be considered 'dangerous goods'. Commercial carriers have shipping regulations—ensure the sample packaging and labelling meet the requirements. Contact the courier company for details prior to sampling.

8.2. Analysis of samples

Once the microalgae samples have been collected and/or fixed with Lugol's solution the samples can be transported to the analytical laboratory and provided to the phytoplankton expert for identification and counting purposes.

8.3. References and additional reading

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Appendix 1 Equipment checklists

Table 1: Equipment checklist for phytoplankton water samples

Equipment and consumables	✓
Note book or equivalent	
Fresh water for washing skin and hand washing equipment	
Gloves	
Integrated hose-pipe sampler – 5m length of 2.5cm diameter plastic piping with a weighted collar at one end	
Rigid pipe fitted with a one-way valve (if preferred to a flexible hose-pipe)	
Cord (attached to the hose and boat)	
Rubber cork that fits one end of the hose	
5L bucket	
Pole type sampler; sampling pole with attachment to hold sample bottle/container	
200mL and 500mL sample bottles and lids	
Lugol's solution and disposable pipette	
Cool box and ice	

Table 2: Equipment checklist for benthic algae samples

Equipment and consumables	✓
Note book or equivalent	
Fresh water for washing skin and hand washing equipment	
Gloves	
Benthic sampler (e.g. Eckman grab) or a rigid plastic corer (e.g. PVC or polycarbonate pipe)	
Nets or rakes	
Container with a fitted lid or a zip lock bag	
Lugol's solution and disposable pipette	
Cool box and ice	

Table 3: Equipment checklist for marine benthic algae identification

Equipment and consumables	✓
Gloves	
Rake or garden fork	
150mL high density polyethylene screw top jar	

Section C: Biological Assessment

Zip lock bag with absorbent material	
Portable refrigerator	
Cool box and ice	

Appendix 2 Preparation and use of Lugol's solution

To make up Lugol's solution

Chemicals

- Potassium iodide (KI)
- Pure iodine crystals
- Glacial Acetic acid
- Distilled water

Note: Take care when handling, avoid contact with skin because the glacial acetic acid is highly corrosive and Lugol's solution will readily stain skin (brown/orange colour).

Method

1. Mix 20g of potassium iodide (KI) with 180mL distilled water.
2. Dissolve 10g of pure iodine crystals in this solution.
3. Add 20mL glacial acetic acid.
4. Store the solution in a dark coloured glass bottle inside a dark chemical storage cupboard. The bottle should be labelled with:
 - the date of preparation
 - name of the analyst who prepared the solution
 - a use by date.
5. This stock solution should not be used 6 months after the date of preparation.
6. The bottle should be sealed around the cap with Parafilm®.
7. If using polyethylene bottles for water samples, note that these bottles will absorb the iodine in Lugol's solution very quickly, and should not be used for long term storage. Staining also occurs when numerous water bottles (containing Lugol's fixed samples) are stored in a plastic container (i.e. an esky). If Lugol's fixed water samples are to be stored prior to concentration by settling or centrifugation in the laboratory, they should be stored in amber glass bottles in the dark and processed as soon as possible.

Note: The iodine in Lugol's solution not only preserves and stains the phytoplankton but it also increases their density making them heavier so that they sink more readily to the bottom of a settling cylinder.

Guidance on seagrass monitoring

Version: February 2018

9. Guidance on seagrass monitoring

9.1. Purpose and scope

This document provides background information on seagrass monitoring and where to get advice on monitoring approach and relevant techniques.

9.2. Associated documents

Biological assessment: Guidance on using Photosynthetically Active Radiation (PAR) as a method to measure light availability for photosynthetic organisms facing acute impacts.

9.3. Introduction

Seagrasses can be an excellent indicator of environmental change and impact, as well as being an incredibly valuable coastal habitat performing a range of critical ecosystem functions, such as stabilising bottom sediments, and providing fisheries habitat and food for dugong and turtles (Seagrass-Watch HQ, 2006-2015). However, deciding how and what to monitor is not straight forward. In Queensland, seagrasses include a variety of species, occur in a range of landscapes from sparse through to continuous cover, and across a broad range of depth gradients from shallow intertidal (**Figure 93**) through to depths greater than 60m in the Great Barrier Reef lagoon. As a consequence not all areas of “seagrass” behave the same, even under natural conditions; some meadows remaining relatively stable through to those that are naturally highly variable. There are also 15 different species of seagrass in Queensland waters, all of which have different lifecycles and ecological requirements. Taking a “one size fits all” approach to monitoring is unlikely to yield desired outcomes and any seagrass monitoring program should be designed with careful consideration of the nature and location of the meadow, the species involved and the questions that needs to be answered.



Figure 93: A seagrass meadow found on tidal flats

9.4. Scale of monitoring

A critical consideration is the scale at which monitoring should be conducted. For example, small-scale permanent transect approaches are most suited to answering questions related to local issues, such as assessments of impacts from point sources or assessments of discrete impacts to specific sensitive receptor sites. However, many seagrass landscapes are highly dynamic and present a shifting mosaic of biomass hot spots within their boundaries, as well as substantial shifts in the spatial footprints of the meadows (Seagrass-Watch HQ, 2006-2015). Therefore, questions regarding the overall state of seagrasses in a particular bay or region are best addressed using methods that integrate much greater areas that are considered representative; e.g. using multiple sites within a meadow and multiple meadows within a bay or region. In Queensland, there are excellent examples of both kinds

of approaches that have been conducted over long time-frames (Coles et al. 2015). These programs present standardised approaches and adopting these methods at new sites has the distinct advantage of allowing for easy regional and state-wide comparisons to be made, thereby contextualising the changes observed locally.

9.5. Variables to be measured

Another important consideration is the variables that should be assessed. There are a large number of different seagrass condition variables that could be collected, ranging from the molecular scale all the way through to whole plant and landscape changes. Again, the questions being asked of the monitoring program should dictate the most suitable variables for monitoring. However, if tracking the change in seagrass is the overarching goal, then there are some fundamental variables that should be incorporated:

- Change in seagrass meadow area
- Change in biomass or cover within those areas
- Significant shifts in species composition
- Changes in sediment characteristics and topography
- Light (measured as photosynthetically active radiation (PAR²⁰)).

In addition to these, there are an enormous range of other important factors that could be included in monitoring, including measurements of seed-banks and reproductive output, assessment of nutrient status, plant carbohydrate stores, herbivory, epiphyte cover, changes in gene expression indicating stress and assessment of key controlling variables such as light and temperature. The final mix of monitoring variables will depend on the resources available, logistical constraints and what the monitoring is trying to achieve.

9.6. Frequency of monitoring

Seagrasses in Queensland are highly seasonal, so the timing of monitoring is critically important. In part the frequency of monitoring will depend on what the monitoring objectives are. An annual assessment of seagrass condition could reasonably be performed with one sampling event conducted during the peak season for seagrass abundance (typically between September and December). This is important as some species are annuals and present only as a seed bank through winter. However, if tracking seasonal change is important to the program, then more frequent (ideally at least quarterly) sampling is required. An even higher frequency of sampling may be needed if the monitoring is intended to assess compliance or impacts associated with a particular development or discharge event. The variables selected for measurement in the monitoring program will also have a bearing on the timing and frequency of sampling. For example, flowering and sexual reproduction for some species can be a highly variable and occur over relatively short timeframes, and reliable data may require multiple sampling events during the reproductive seasons to ensure they are adequately assessed.

9.7. Establishing appropriate baselines

Monitoring programs will generally be required to assess change from some baseline or reference condition. The highly seasonal nature and potentially large inter-annual changes that can occur in Queensland's seagrasses mean that the longer the baseline period of data collection is, the better. Often this is not possible with programs triggered by a specific event, but a minimum of three to five years is likely to provide a useful context and allow some level of assessment of the degree of inter or intra-annual change, and thereby place future monitoring results in context. Recent work has suggested that even longer timeframes may be required to set baseline conditions for detailed seagrass condition reporting (see Gladstone Healthy Harbour Program²¹ report card for example).

9.8. How to decide what monitoring approach to take

To decide on the best monitoring approach for a given situation it is recommended that one of the specialist seagrass monitoring groups in Queensland is consulted. Some links are provided below. The two major ongoing seagrass monitoring programs for Queensland are based out of James Cook University:

Monitoring in high risk areas of the state as part of the James Cook University ports seagrass monitoring. See: www.jcu.edu.au/portseagrassqld

²⁰ Light requirements (specified as PAR) have been added as a WQO for some coastal waters containing seagrasses under the Environmental Protection Policy (Water).

²¹ <http://ghhp.org.au/>

Seagrass-Watch. See: <http://www.seagrasswatch.org/home.html>

Some monitoring techniques are provided within these links. The Great Barrier Reef (GBR) Marine Monitoring Program is also a valuable source of information for Queensland waters (www.gbrmpa.gov.au/managing-the-reef/how-the-reefs-managed/reef-2050-marine-monitoring-program) and global programs such as SeagrassNet (<http://www.seagrassnet.org/global-monitoring>) have comprehensive manuals to download for monitoring that can be used as guidance.

Other useful web pages for seagrass monitoring in Queensland

James Cook University Seagrass Ecology Group: www.segrasseecology.com

Griffith University Australian Rivers Institute: <https://www.griffith.edu.au/environment-planning-architecture/australian-rivers-institute>

University of Queensland Remote Sensing Research Centre: <https://www.gpem.uq.edu.au/rsrc>

9.9. References and additional reading

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Guidance on using Photosynthetically Active Radiation (PAR) as a method to measure light availability for aquatic photosynthetic organisms facing acute impacts

Version: February 2018

10. Guidance on using Photosynthetically Active Radiation (PAR) as a method to measure light availability for aquatic photosynthetic organisms facing acute impacts

10.1. Purpose and scope

This purpose of this document is to provide general guidance in the use of photosynthetically active radiation (PAR) sensors for monitoring aquatic photosynthetic organisms such as seagrasses.

Light requirements (specified as PAR) are included as a Water Quality Objective (WQO) for some coastal waters under the Environmental Protection (Water) Policy 2009.

10.2. Associated documents

Sampling design and preparation:

Record keeping, including taking field photographs and videos

Sampling scope and design

Permits and approvals

Biological assessment: Guidance on seagrass monitoring

10.3. Introduction

Photosynthetically active radiation (PAR) refers to the spectral range (400 to 700 nanometres) of solar radiation that is used in photosynthesis. This is relevant for primary producers such as seagrass and phytoplankton as well as for most reef-building corals which contain photosynthetic algae (zooxanthellae) that live in their tissues and provide coral with their primary food source. Insufficient PAR can lead to reduced growth or loss of seagrass, corals and other photosynthetic organisms. Measurement of PAR can therefore be used as a surrogate indicator to assess potential impacts on photosynthetic plants. Telemetered PAR data allows for real time monitoring and management of activities that can potentially impact these organisms, such as dredging. PAR is preferred to turbidity for measuring potential impacts from total suspended solids to seagrass as it provides a biologically relevant indicator.

Acute related pressures or impacts are defined as typically less than 3 months and related to either discrete coastal development activities or weather-related events that impact water quality (such as cyclones) (Collier et al. 2016). However, the specific time-scale over which an acute event occurs can be longer. For instance, a 12-month dredging campaign may be classified as an acute pressure since it is a well-defined activity that adds pressure to the light environment that is distinct from background water quality conditions. Chronic pressures would relate to longer-term changes to the light environment such as those that occur due to changes in river catchment activities and sediment loads from coastal communities. These are sustained in the long term without a clearly defined end date for the pressure.

10.4. Planning a monitoring program

For field deployment, loggers should be attached to permanent station markers. For intertidal and subtidal areas, this is above the sediment-water interface such that the sensor is at the same height as the species being monitored (e.g. top of the seagrass canopy), so they are collecting light equivalent to what is reaching benthic flora. When designing a monitoring program, key considerations are:

The most commonly used sensors, and the ones recommended for monitoring, are flat “2π” sensors. Differences in sensor design and shape will influence how they intercept radiation – different instruments may produce different measurements (Long et al 2012 and references therein); therefore, it is recommended that the same brand of sensor be used for all sites in a monitoring program.

PAR should be recorded at or just above the flora that are being managed because PAR changes with depth due

to light attenuation,

A reference logger that measures incident surface PAR should be deployed near the site (in the air) as benthic PAR is in part a function of the incident PAR reaching the water surface (which can be impacted by cloud cover etc.), so it is needed to separate out water column and atmospheric effects.

PAR should be logged continuously and recorded, at a minimum, of 15 minute intervals.

Two loggers should be deployed at all sites, in case of loss or logger fouling.

It is essential that a self-cleaning or wiper system is used with the loggers to ensure the sensor remains clean (un-fouled) – typically the lens is wiped every 15 minutes.

If real time management of an activity is required, these instruments should be combined with a telemetry system.

Each light logger has a unique serial number that should be recorded within a central secure database. The logger number should be recorded on the monitoring site datasheet with the time of deployment and retrieval.

At permanently submerged sites, the loggers should be checked by SCUBA divers every six to twelve weeks and replaced if fouled. The length of time between logger replacements is site-specific due to variation in fouling rates.

Photographs of the light sensor and/or notes on the condition of the sensor should be recorded when the logger is retrieved. If major fouling is noted (e.g. wiper failure), the data are truncated to include only those data collected before fouling began. If minor fouling is noted (up to ~25% of the sensor covered), back corrections can be made to the data, allowing for a linear rate of fouling (linear because with minor fouling it is assumed that the wiper was retarding algal growth rates, but not fully inhibiting them).

Loggers must be calibrated or checked against a certified reference Photosynthetically Active Radiation (PAR) sensor using a stable light source enclosed in a casing that holds both the sensor and light source at a constant distance. Calibration must be repeated between each deployment period.

Manufacturer's instructions must be followed at all times.

A correction factor should be applied to logged data to account for the difference between the calibration (if performed in air) versus the deployment conditions (in water). This is due to the difference in light absorption properties between air and water by the sensor (Kirk 1994). When assessing data from intertidal loggers, the application of the correction factor must take into account exposure history (i.e. shifts between inundation and exposure to air), and only be applied to data when the logger was inundated, assuming the calibration was done in air.

10.5. Assessment of data

Light is measured as instantaneous irradiance ($\mu\text{mol m}^{-2} \text{s}^{-1}$). Accumulation of instantaneous irradiance should be calculated as daily PAR or irradiance (I_d , $\text{mol m}^{-2} \text{d}^{-1}$), which is standard practice and a useful way of expressing the PAR data for management of photosynthetic benthic light.

A simple close approximation to convert a daily series of regularly measured instantaneous irradiance readings ($\mu\text{mol m}^{-2} \text{s}^{-1}$) to daily light integrated PAR or irradiance (I_d , $\text{mol m}^{-2} \text{d}^{-1}$) is as follows:

Assess recording intervals, and number of days.

Delete incomplete days, and calculate how many records to expect in total.

Check for missing values. If there are missing values, interpolate instantaneous irradiance readings to fill the gaps.

Check that night time readings are close to zero, and noon readings are within the range of expected values ($\sim 2000 \mu\text{mol m}^{-2} \text{s}^{-1}$) for surface readings when sunny; ideally, all instruments will cross-calibrated the loggers, and deployed side by side).

Apply the following formula:

$$\text{Daily PAR } I_d \text{ [mol m}^{-2} \text{ d}^{-1}] = (\text{sum of all instantaneous values}) \times 24 \times 60 \times 60 / 1,000,000 / N \times D$$

$$= (\text{sum of all instantaneous values}) \times 0.0864 / N \times D$$

Where N is the number of samples taken in total, and D is the number of complete days of logger deployment.

A logger was programmed to record one instantaneous value every 15 min for a day. To calculate the Daily PAR:

$$N = 60 \times 24 / 15 = 96.$$

Sum of the 96 instantaneous readings = 2119.47 $\mu\text{mol m}^{-2} \text{s}^{-1}$.

$D = 1$.

Thus, Daily Par (I_d) = 1.9075 $\text{mol m}^{-2} \text{d}^{-1}$ at this location.

Regional catchment-level Water Quality Objectives are being implemented under Queensland's Environmental Protection (Water) Policy 2009 for Great Barrier Reef catchments. Readers should refer to the specific basin for prescribed Water Quality Objectives for the protection of seagrasses. Generally guidelines in coastal waters are specified as:

over a rolling seven day average for deep (>10 m) water

over a rolling 14 day average for shallow inshore (<10 m) water.

Note: Absolute light requirements for seagrass may vary between sites and species. Values described in the Environmental Protection (Water) Policy 2009 provide a conservative guide to the levels of light likely to support seagrass growth from acute water quality impacts. Locally derived, absolute, and species-specific thresholds ideally should be obtained for management of specific activities likely to impact on the light environment. Higher light requirements may be needed for the management of longer term chronic impacts.

10.6. References and additional reading

Chartrand KM, Ralph PJ, Petrou K and Rasheed MA. (2012) Development of a Light-Based Seagrass Management Approach for the Gladstone Western Basin Dredging Program. DAFF Publication. Fisheries Queensland, Cairns 126 pp.

Collier, CJ, Chartrand K, Honchin C, Fletcher A and Rasheed, M 2016, Light thresholds for seagrasses of the GBR: a synthesis and guiding document. Including knowledge gaps and future priorities. Report to the National Environmental Science Programme. Reef and Rainforest Research Centre Limited, Cairns (41pp.).

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Direct toxicity assessments

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11. Direct toxicity assessments

11.1. Purpose and scope

This document provides general advice on requirements and considerations for sample collection for direct toxicity assessments (DTA).

11.2. Associated documents

Physical and chemical assessment:

- *Manual collection of surface water samples (including field filtration)*
- *Collection and preservation of sediment*

Sampling design and preparation:

- *Permits and approvals*
- *Record keeping, including taking field photographs and videos*

11.3. Introduction

Direct toxicity assessments (DTA) involve the use of toxicity tests to determine the acute and/or chronic toxicity of waste water discharges or total pollutant loads in receiving waters. The use of DTA allows an assessment of the toxicity of mixtures of chemicals rather than individual chemicals, and is part of both the ANZECC and ARMCANZ (2000) (water) and Simpson et al. (2013) (sediment) decision frameworks for environmental protection.

The specialised nature of these bioassays (toxicity tests) requires specific expertise rarely available outside of dedicated testing facilities. Most DTA programs rely on the availability of off-the-shelf toxicity tests that utilise standard test species. These standard tests are often accredited for use by the National Association of Testing Authorities (NATA), which provides confidence in the test results. However, the use of site-specific species should be considered where standard species are deemed unrepresentative of the local ecosystem, and can be used provided standard toxicity testing procedures are adhered to. Advice on the use of local test species should be obtained from the testing laboratory.

DTA can be conducted on water, whole sediments or aqueous extracts (e.g. elutriate or pore-waters) from sediments. Wastewater and sediment samples destined for DTA often need to be collected by non-specialists. When collecting samples for DTA, there will generally be a requirement to collect:

- wastewater from a specific release point (or sediment from a downstream location)
- uncontaminated water to be used as a diluent in toxicity tests.

11.4. Preparation for sampling

The analysing laboratory should be contacted early on in the planning phase before sample collection to obtain detailed advice on the appropriate procedures for collecting water or sediment samples for DTA.

When collecting sediment, wastewater or bulk diluent water for the purpose of conducting a DTA, it is important to collect the sample according to the testing laboratory's advice, which should include:

- volume of wastewater, diluent or sediment required. The volume of each of these will depend upon the type of testing (e.g. larger organisms will require more sample volume), and the number of tests to be conducted.
- type of sample bottle/container. These may be provided by the lab. If not, the lab will advise on the requirements for sample containers
- field storage requirements (i.e. immediately placed into esky with ice bricks)
- instruction on any additives for the purpose of preservation or stabilisation
- details on laboratory holding times for the samples
- instruction on sample security
- instructions for transport
- instructions for mandatory in situ water quality measurements.

11.5. Sampling considerations

11.5.1. Waters

Wastewater samples collected for the purpose of DTA should represent the variation in water quality as the concentration of contaminants in wastewaters can vary over time (i.e. minutes, hours or days), and space (i.e. across a channel profile). It is important to:

- define the sampling strategy that will sample the expected variability or
- select for worst-case water quality.

Where wastewater is well mixed and homogenous over time, a single grab sample is likely to be appropriate. However, where a wastewater is expected to vary over time, then a composite sample may be appropriate.

Note: Although composite samples may be advantageous in some scenarios, composite samples may not provide information on the maximum concentration of contaminants which may be of interest in a DTA.

Follow the laboratory instructions for requirements for sampling wastewater for DTA analysis. For the actual collection of the wastewater, follow the steps provided in the Manual collection of surface water samples (including field filtration) document.

Additional requirements specific to collecting wastewater for DTA analysis include:

- Collect wastewater from a specific release point. Wastewater needs to be collected directly from a water off-take or at the end-of-pipe prior to mixing with any receiving environment waters. Ensure GPS co-ordinates are taken and noted.
- Wastewater samples collected for the purpose of DTA are typically not filtered prior to storage for transport, unless specifically required by the analyst.
- Water samples must be delivered to the analyst within the prescribed holding time – this is generally within 24 hours to limit the degree of sample degradation.

Note: Delay in the delivery of water samples to the analyst can also result in significant losses or transformations of toxicants (e.g. chlorine, ammonia, cyanide, pesticides), which need to be considered when interpreting the results of DTA. This is particularly important where the time for transport exceeds maximum holding times for those analytes. Therefore, where feasible, all relevant indicators need to be measured at the site and time of collection. Alternatively, additional water samples need to be collected and preserved in an appropriate manner for later analysis. Seek advice from the DTA analyst who can assess this requirement on a case-by-case basis.

It is important that physicochemical parameters of both the wastewater and the receiving environment are measured in situ (wherever possible), and then again prior to conducting any DTA in the testing laboratory (by laboratory staff).

The physicochemical parameters include:

- temperature
- pH
- electrical conductivity
- dissolved oxygen
- turbidity.

11.5.2. Diluent water

Toxicity testing requires the use of a series of different wastewater concentrations. This is achieved by dilution of the wastewater sample with water. Generally speaking, a dilution series is prepared using water collected from a location in the receiving waters known to be uncontaminated by the wastewater being investigated (i.e. upstream, up-current, or local control site). Ensure GPS co-ordinates are taken and noted.

Follow the laboratory instructions for requirements for collecting diluent water. For the actual collection of diluent water, follow the steps provided in the Manual collection of surface water samples (including field filtration) document.

Additional requirements specific to collecting diluent water for DTA analysis include:

A large volume of diluent water (uncontaminated water) is required for DTA

It is important that samples are collected up-current of significant in-stream structures and known point sources of pollutants, except where the contribution of these pollutants are to be included as a component of the toxicity tests.

- Water sample containers must be filled to the top, leaving no airspace.

- Water samples are not filtered unless required by the analyst.
- Water samples must be delivered to the analyst within the prescribed holding time – this is generally within 24 hours to limit the degree of sample degradation.

11.5.3. Sediment

Sediment samples are typically more heterogeneous (poorly mixed) than water and wastewater samples. In order to collect a representative sample consideration needs to be given to the exact location for collection, and whether a composite sample from a wide area, or multiple sub-samples, is more appropriate than a single discrete sample. Where multiple sub-samples are taken to produce a composite sample, or taken as discrete samples in preference to a composite sample, at least three sub-samples should be used to provide adequate replication.

Follow the laboratory instructions for requirements for collecting sediment (i.e. volume, bottle type, holding times etc.). For the actual collection of sediment, follow the steps provided in the Collection and preservation of sediment document.

Additional requirements specific to collecting sediment for DTA analysis include:

- A record of the number of sub-samples used in the creation of a composite samples and the depth at which they are taken must be standardised between sites and defined prior to collection.
- The distances between sub-sampling sites must be standardised between sites and defined prior to collection.
- Sediment samples must be delivered to the analyst within the prescribed holding time. Holding times will vary depending on how the sediment is to be used.
 - Where pore-waters are to be extracted for DTA, holding times should be as short as possible (i.e. 24 hours).
 - Where whole sediments are to be used for DTA, holding times and sample storage conditions should meet the requirements described for the specific compounds present, but generally no longer than two weeks (with appropriate refrigeration; <6°C). Seek advice from the analysing laboratory.

It is important that physicochemical parameters of sediment, and the receiving environment are measured in situ (wherever possible), and then again prior to conducting any DTA in the testing laboratory (by laboratory staff).

The physicochemical parameters include:

- pH
- electrical conductivity
- reduction/oxidation (REDOX) potential

Note: Disturbance of sediments invariably results in changes to various other chemical equilibriums (i.e. ammonia/ammonium, sulphide speciation, metal speciation), which need to be considered when interpreting the results of DTA. Additional sediment samples may need to be collected and preserved in an appropriate manner for later analysis.

11.6. References and additional reading

ANZECC & ARMCANZ 2000, Australian and New Zealand Guidelines for Fresh and Marine Water Quality, National water quality management strategy, Paper 4, Australian and New Zealand Environment and Conservation Council and Agriculture and Resource Management Council of Australia and New Zealand, Canberra, ACT.

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Section D: Data Handling

Custodianship and management

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1. Custodianship and management

1.1. Purpose and scope

The purpose of this document is to provide information on data handling and the importance of custodianship and management.

1.2. Importance of data custodianship and management

Reliable and defined custodianship and data management is essential to ensure data are collected, maintained and used appropriately. Good custodianship of data provides accountability for datasets and gives the user confidence in the level of integrity, timelines, precision and completeness of datasets.

1.3. What are data?

Data can be measurements or statistics obtained from measuring devices or observations, usually presented in a numerical or structured format.

A collection of data may be referred to as a dataset, often held electronically in files or a database.

Generic classification of data includes:

Time series data which are a set of observations, results, or other data obtained over a period of time from consistent or known monitoring locations, often at regular intervals.

Spatial data which are data that refer to specific geographic areas. Such records would generally include a geographic reference, e.g. map references, latitude and longitude references, river catchment areas, local government areas etc.

Metadata which are datasets that detail the content, quality, condition, and other relevant characteristics of the data. Metadata often describes any data that are processed, organised or classified into categories, images, graphs, etc. for a designated purpose. If data has been modified at all (i.e. outliers removed) this must be captured in the metadata.

1.4. Data custodians

All data, whether generated by government or by organisations external to government, must be managed by a custodian.

A data custodian can be defined as a person or organisation that is responsible for ensuring data are collected, maintained and made available according to standards, policies or other licences, agreements or specifications. In the case of regulatory work undertaken by organisations external to government, the data custodian would typically be the proponent or the approval holder. A custodian can act as a contact with the regulatory authority to allow the release or use of data by other parties²².

Custodians are responsible for ensuring that the following minimum standards are applied to each dataset:

The method and process for data collection must be clearly documented.

Data are fully validated and quality assured with sufficient detailed metadata to enable the use by third parties without referring to the person or organisation who collected the data.

²² For Queensland Government custodians, public release of data is governed by the publishing standards classifications in line with the Queensland Government Information Security Classification Framework (QGISC) need to be obtained.
<https://www.qgcio.qld.gov.au/products/qgea-documents/549-information-security/2417-queensland-government-information-security-classification-framework>

Ownership, access constraints and licence conditions are associated with any recorded data.

1.5. Data quality assurance and quality control

Data custodians must ensure as far as practicable, the accuracy, currency and timeliness of data supplied. Data quality assurance and quality control minimises any potential problems that may arise with data use by preventing errors and reviewing data management practices. Good data management practices include:

- the provision of clear documentation
- the use of standard definitions and classifications
- the maintenance of metadata (including quality attributes of the data)
- the appropriate storage of the data itself.

Some public sector examples of documented standards and guidelines for water data management and publication include:

resource and publication guides for publishing datasets to the Queensland Government Data Portal, including a resource formatting and metadata guide

the Draft National Industry Guideline for water quality metadata for water quality data collected and supplied to the Australian Government Bureau of Meteorology under the Water Regulations 2008.

Organisations that use data for regulatory purposes, including those submitting data to the Queensland Government, are encouraged to have a structured data management system that provides reliable and secure consistent storage, access and reporting.

Specific quality assurance protocols and procedures are generally required for continuous monitoring techniques where higher frequency data is collected over a period of time. In such cases, instrument measurements can “drift” away from real values with time and it may be necessary to adjust the time series data to account for such changes. The application of any data corrections must be based on documented procedures and be defensible. Erroneous data should be removed from the dataset before use, particularly where data is to be submitted to the Queensland Government for regulatory purposes.

1.6. Regulatory purposes

There are a number of situations where individuals and organisations may need to submit water monitoring information (and related metadata) to the Queensland Government for the purpose of regulatory decision making. This could include, but is not limited to, purposes such as:

- Environmental Impact Statements
- Environmental Approvals and related conditions
- Environmental Management Plans
- Environmental Evaluations.

Any water monitoring data provided to the Queensland Government should be collected in accordance with this document and other relevant standards, guidelines and policies. In many cases, water monitoring data is required to be submitted electronically. As a minimum, data submitted should include:

- a unique sample identifier
- information about the sampling location identifier
- sampling location description
- sampling date
- sampling time
- details of the person who took the sample (name and contact)
- constituent measured
- units
- qualifiers (such as equal to or greater/less than signs)
- results, method used to derive the result and the names of organisation/s responsible for sample collections and field or laboratory analysis.

Supporting information on sampling locations is essential and should include:

unique sampling location identifier

sampling location descriptions

latitudes and longitudes

the datum used, and relevant stream/basin names where applicable in addition to maps or diagrams showing the monitoring point locations.

The data must be supplied to the Queensland Government in:

the specified electronic file format and/or template where requested.

Portable document formats (pdfs) are not considered an acceptable file format for water monitoring data submission.

Acceptable file formats may include excel, text or comma separated value (csv) files, depending on the application.

Information on the methods used for monitoring, including both sampling and analysis, should be kept and supplied. Data custodians submitting data must have sufficient records to demonstrate that any sampling and analysis has been undertaken in accordance with this manual.

The provision of correct and accurate data is the sole responsibility of the data custodian and the Queensland Government will not be held responsible for incorrect data submitted by other organisations or agencies. The Queensland Government also reserves the right to use monitoring data that has been provided to it by any organisation for any purpose it sees fit including supply of data to a third party.

Note: For more information on water data standards, including metadata, refer to Australian Government Bureau of Meteorology, Water Quality Metadata Guideline.

1.7. References and additional reading

Queensland Government. Publishing standards for data.qld.gov.au. Available from:
<https://publications.qld.gov.au/dataset/publishing-standards-data-qld-gov-au>

Australian Government Bureau of Meteorology, Water Data Collection Standards. Available from:
<http://www.bom.gov.au/water/standards/datacollection/index.shtml>

Australian Government Bureau of Meteorology, Water Quality Metadata Guideline.
<http://www.bom.gov.au/water/standards/projects/waterqlty.shtml>

http://www.bom.gov.au/water/standards/documents/NI_GL_101.00-2016.pdf

Queensland Government information security classification framework <https://www.qgcio.qld.gov.au/products/qgea-documents/549-information-security/2417-queensland-government-information-security-classification-framework>

Units and concentrations

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2. Units and concentrations

2.1. Commonly used concentration units

Commonly, concentration units are presented using units in the form of mass per volume (e.g. milligrams per litre, mg/L, mg.L⁻¹) for water samples or mass per mass (e.g. milligrams per kilogram, mg/kg, mg.kg⁻¹) for sediment, soil or biota samples. However, units expressed as parts per a number (e.g. parts per million, ppm) may still be encountered.

The conversion of parts per number data to concentration units can be confusing. For this reason, a range of examples are provided in Table 12. Prefixes and multiplication factors used for the conversion of units are presented in Table 13.

Table 12: Conversion of the parts per number units to concentration units. Common concentration units used in environmental science

Parts per number units		Equivalent units mass per mass or mass per volume units				
Parts per thousand	‰	g/kg or g.kg ⁻¹	mg/g or mg.g ⁻¹	g/L or g.L ⁻¹	mg/mL or mg.mL ⁻¹	µg/µL or µg.µL ⁻¹
Parts per million	ppm	mg/kg or mg.kg ⁻¹	µg/g or µg.g ⁻¹	mg/L or mg.L ⁻¹	µg/mL or µg.mL ⁻¹	ng/µL or ng.µL ⁻¹
Parts per billion	ppb	µg/kg or µg.kg ⁻¹	ng/g or ng.g ⁻¹	µg/L or µg.L ⁻¹	ng/mL or ng.mL ⁻¹	pg/µL or pg.µL ⁻¹
Parts per trillion	ppt	ng/kg or ng.kg ⁻¹	pg/g or pg.g ⁻¹	ng/L or ng.L ⁻¹	pg/mL or pg.mL ⁻¹	fg/µL or fg.µL ⁻¹

2.2. Nutrient concentration conversions

Results from nutrient analyses can be reported in two ways – as the whole compound or as the principal element in the compound. For example, nitrate may be reported as nitrate (NO₃) or nitrate as nitrogen (NO₃-N). When assessing results against guidelines and standards, or when comparing data from different sources, it is important to compare like with like and convert the results if needed.

Table 13: Commonly used unit prefixes

Prefix	Symbol	Multiplication factor	Example
giga	G	10 ⁹	Gigalitre (GL) = 1 x 10 ⁹ L
mega	M	10 ⁶	Megalitre (ML) = 1 x 10 ⁶ L
kilo	k	10 ³	Kilometre (km) = 1 x 10 ³ m
deci	d	10 ⁻¹	Decimetre (dm) = 1 x 10 ⁻¹ m
centi	c	10 ⁻²	Centimetre (cm) = 1 x 10 ⁻² m

milli	m	10 ⁻³	Millilitre (mL) = 1 x 10 ⁻³ L
micro	μ	10 ⁻⁶	Microgram (μg) = 1 x 10 ⁻⁶ g
nano	n	10 ⁻⁹	Nanogram (ng) = 1 x 10 ⁻⁹ g
pico	p	10 ⁻¹²	Picogram (pg) = 1 x 10 ⁻¹² g
femto	f	10 ⁻¹⁵	Femptogram (fg) = 1 x 10 ⁻¹⁵ g

Example of conversions:

1 mg/L of nitrate NO₃-N = 4.43mg/L NO₃

To convert mg/L NO₃-N to mg/L of NO₃ multiply result by 4.43

To convert mg/L NO₃ to mg/L NO₃-N divide by 4.43.

Other conversion factors:

1.00 mg/L of nitrite as N (NO₂-N) = 3.28mg/L nitrite (NO₂)

1.00 mg/L of ammonia (NH₃) as nitrogen (N) = 1.22 mg/L of ammonia as ammonia

1.00 mg/L of ammonium (NH₄) as nitrogen (N) = 1.29mg/L of ammonium as ammonium

1.000 mg/L of phosphate (PO₄) as phosphorus (P) = 3.066mg/L of phosphate as phosphate.

2.3. Conductivity units and abbreviations

The unit of measurement for conductivity is siemens (S) per unit of length of water that the current is passed through. Some common electrical conductivity (EC) unit expressions used for reporting conductivity are:

microsiemens per centimetre (μS/cm or μS.cm⁻¹)

millisiemens per centimetre (mS/cm or mS.cm⁻¹)

decisiemens per metre (dS/m or dS.m⁻¹)

millisiemens per metre (mS/m or mS.m⁻¹).

The SI unit is millisiemens per metre (mS/m or mS.m⁻¹). Equivalence relationships among these units include:

1dS/m = 1mS/cm = 100mS/m = 1000μS/cm

1mS/m = 10μS/cm