NIWA
Taihoro Nukurangi

SHMAK

Stream Health Monitoring and Assessment Kit

User Manual

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SHMAK Stream Health Monitoring & Assessment Kit User Manual

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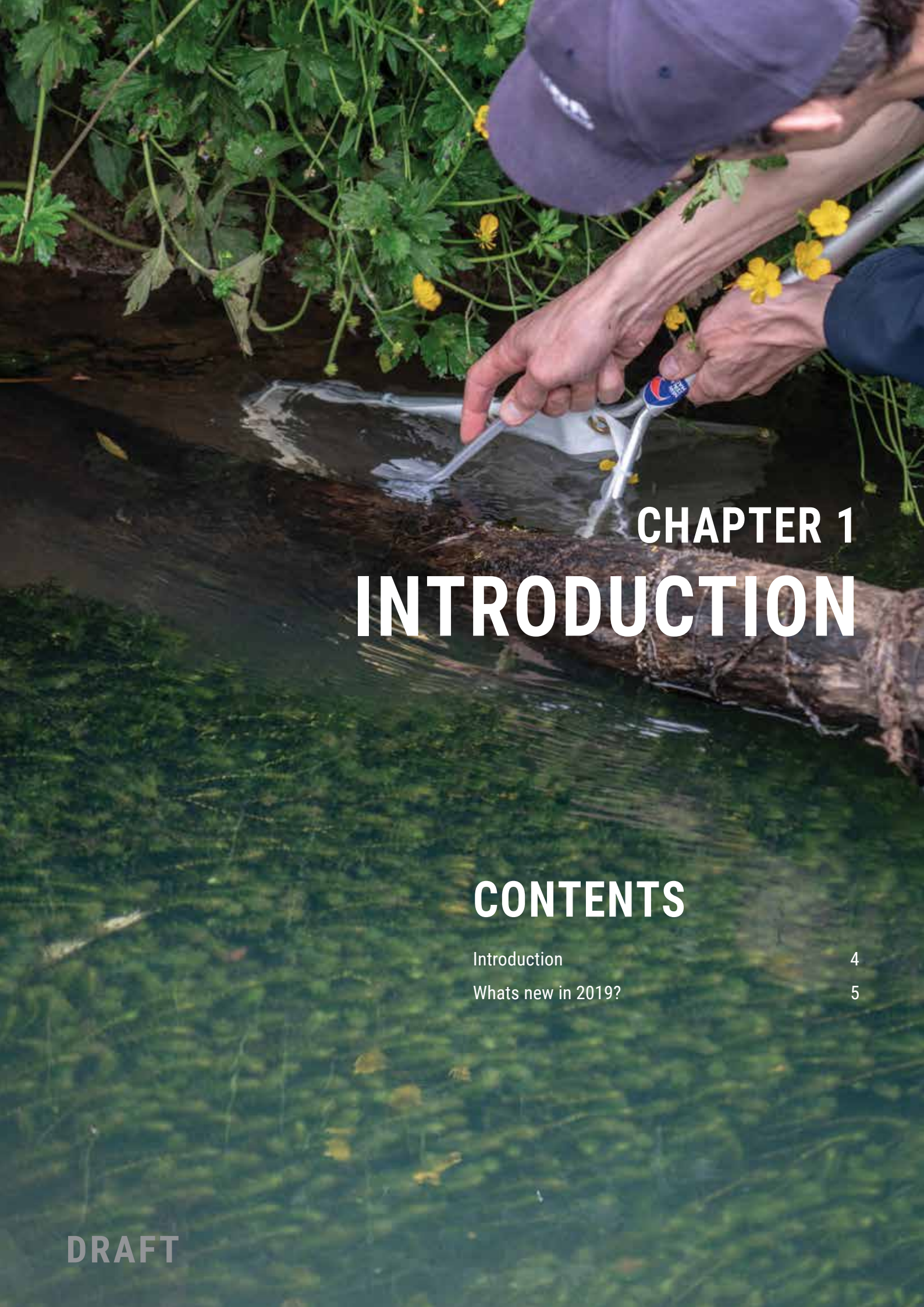

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INTRODUCTION

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INTRODUCTION

For over 20 years, NIWA's Stream Health Monitoring Assessment Kit (SHMAK) has provided land owners, iwi, school and community groups with a simple, scientifically-sound resource to monitor the ecological health of New Zealand's streams.

First released in 1998, SHMAK was developed as a joint project between Federated Farmers and NIWA. The kit and user guide were supported by many organisations and local landcare groups, with funding provided from the Ministry for the Environment's Sustainable Management Fund. A few years later the New Zealand Landcare Trust came on board and, together with continued funding from the Ministry for the Environment, contributed to education and promotion of SHMAK. Additional support was provided by the Kaupapa Taiao Unit of Te Rūnanga o Ngāi Tahu, culminating in the release of Version 2K – A Tool for Kaitiaki in 2002. This version of SHMAK included a strong focus on the principles of Māori scientific research and recognise the pivotal role of iwi in mana whenua, mana moana and kaitiakitanga.

In 2017, a growing interest in New Zealand's fresh water resources, advances in technology – particularly with respect to water quality testing – and a better knowledge of what user groups want and need from the kit, we began revising SHMAK. The kit you see today reflects the valuable input of many organisations and volunteer groups, including financial support from the Ministry for Business Innovation and Employment (MBIE) Envirolink scheme and several regional and city councils.

WHAT'S NEW IN 2019?

The upgraded SHMAK includes:

- a revised suite of equipment and user guide that provide for a more accurate assessment of visual water clarity and new indicators of stream health, including rubbish, dissolved nutrients, and faecal indicator bacteria
- an accompanying suite of training videos produced with Greater Wellington Regional Council, and
- data entry and management tools via a dedicated new website: NZWaterCitizens.co.nz.

As the kit now includes more indicators of stream health, understanding what, when and how to monitor is a little more complex. We recommend spending the time defining the questions you wish to answer about your stream(s) of interest so that you select the most appropriate indicators and methods.

As SHMAK has increased in its scope and public use, we are releasing this manual in draft format to provide users with an opportunity to trial it and provide feedback over the 2019/20 summer field season. This feedback will be used in finalisation of the manual, in particular, to ensure that the methods and supporting information are easy to understand.

Please send you feedback or questions to shmak@niwa.co.nz by 28 February 2020.

Where to from here?

We also encourage users to enter their data on the NZ Water Citizens website (nzwatercitizens.co.nz) and provide feedback through the user forums and contact form on the website. Alternatively, email: shmak@niwa.co.nz.

Enjoy your summer monitoring!

CHAPTER 2
DESIGNING YOUR MONITORING PROJECTS

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WHERE DO I BEGIN?

To design a successful stream monitoring project you will need to consider the why, who, where, what, when and how of monitoring. This chapter provides a process to help you decide why you want to monitor, who you want to use your data and how to design a monitoring plan that fits your goals and your users' needs.

Designing a monitoring project will involve some back and forth. Work through the questions in this chapter, and when you have identified the stream health indicators you are interested in monitoring, read Chapter 3 to get a deeper understanding of what each indicator can tell you. A site visit may show you that some of the indicators you considered are not possible to monitor at your site and you may need to rethink your monitoring plan.

You can build your monitoring project by considering the following:

- Why do you want to monitor? Define your monitoring goals.
- What sort of background information is available for your catchment?
- Where, what, when and how will you monitor? This is the core of your monitoring plan.
- How will you document your monitoring activities and keep track of your monitoring schedule?
- What quality procedures and checks do you need so that your data can be used as you hope?

DEFINING YOUR MONITORING GOALS

Probably the most important step in developing a monitoring project is clearly describing why you want to monitor. Knowing your goals helps you plan all the other steps.

Why do you want to monitor?

Your reasons for starting a volunteer monitoring project might be to:

- **Educate and raise awareness.** To introduce a group to stream ecology or monitoring methods, so people discover the wonders of freshwater ecosystems and learn how to protect them.
- **Describe the current state.** To compare your stream with stream health standards or guidelines. This type of monitoring gives you baseline data for comparing to future changes, and can help to identify potential problems for further study.
- **Assess an impact.** To find out whether land-use activities (e.g., forestry, pastoral farming, horticulture, urban development) or a point source of pollution (e.g., sewage treatment facility) are having an impact on stream health. This might include assessing the effect of your own farm on a stream that flows through your property.
- **Investigate an issue of concern.** To find the cause of a known problem in a stream (e.g., lots of algae).
- **Evaluate a restoration project.** To assess the effect of activities (e.g., riparian fencing or planting) on stream health.
- **Detect trends.** To know whether stream condition is improving or getting worse over time, in response to changes in land use, land or riparian management, other activities (e.g., riparian fencing or planting) on stream health.
- **Contribute to regional monitoring and scientific research.** Volunteer data, especially when collected carefully and consistently through time, may be able to supplement data collected by councils and research organisations.

You might have more than one reason for monitoring. However, it is important to identify no more than three top reasons and develop the project around those. Your goals may change over time as you gain more information about your stream. Revisit the original questions you wanted to answer. Ask yourself if the background information answered your question(s), or changed the question(s) you want answered.

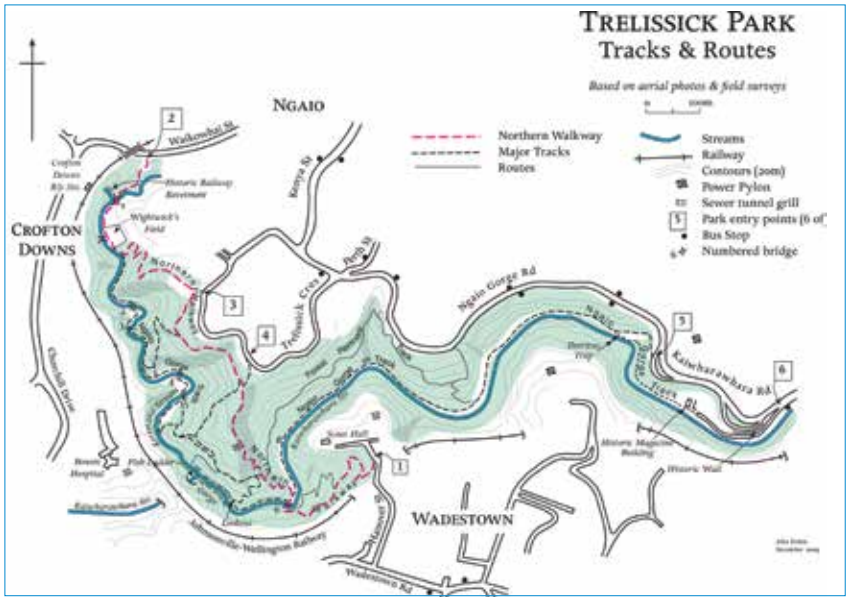
Who will use the monitoring results, and for what purpose?

You might intend your data for yourself only, your students, your streamcare group, your local community (neighbours, catchment group, iwi/hapū), your regional council, an industry organisation (e.g. Beef + Lamb NZ, DairyNZ) or the New Zealand public.

You might intend the data to be used for:

- Education only
- A quick assessment to highlight places needing more detailed investigation
- Monitoring the effects of your own (or your community's) land management
- Adding data to council State of Environment monitoring
- Influencing decision-makers (e.g. local industries, water managers, government).

Your answers to these questions will determine how much training is needed before you monitor, which method you use (for indicators that have a quick and an advanced/detailed option), and what level of data quality and data assurance you need.



GATHER BACKGROUND INFORMATION

Before you develop your monitoring project, you should take time to review the environmental information available for your stream, its catchment (the area of land where its water comes from), and (if relevant) the issue you are concerned about. It is important to gather information about the whole catchment, especially upstream of your site(s). Remember that historical as well as present-day issues may be having an effect on your stream. If you want to monitor impacts of future developments, you need information on the developments too.

Begin by writing down what you and your group already know, making notes on a map where relevant. Then ask what else you need to know.

Sources of information

You can find a lot of information about your stream and catchment from your regional council. Council staff may be able to provide you with maps showing land use, possible pollution sources, and other features that may affect water quality and stream ecological health. Regional councils and NIWA also have up-to-date monitoring data on water quality, aquatic life, streamflow and rainfall from hundreds of sites throughout New Zealand. Data from nearby stream flow (hydrometric) and rainfall monitoring sites can be particularly helpful for interpreting your monitoring results.

Make a catchment map

Mark the boundaries and basic features of your catchment using a topographic map or any other available maps or aerial photos. A topographic map shows the shape and elevation of land forms with contour lines, normally at a scale of 1:50,000. You can find maps and aerial photos on-line or by calling your local or regional council. The aerial photos on www.nzwatercitizens.co.nz, which have REC (River Environment Classification) rivers marked, can also be used.

On the map, mark

- the stream network (including tributaries)
- relevant natural features (e.g. soil types, waterfalls, wetlands)
- land uses and human activities that may affect your stream
- ecologically, recreationally or culturally important places
- existing monitoring sites.

If you are mainly interested in what is happening on a particular parcel of land, it could be useful to make a more detailed map of that area.

For example, for a farm map, mark:

- all streams, including drainage ditches where water flows for at least part of each year
- all buildings (name them, e.g. house, hay shed, milking shed)
- roads and tracks
- water wells
- paddocks (e.g. sheep grazing, cattle grazing, crops, rotated)
- forest blocks (permanent or for harvest)
- areas that flood and areas that are permanently wet.

On the streams, mark:

- regular stock crossings
- vehicle fords
- reaches where stock have access to the water
- any locations where water is taken from the stream
- points on each stream receiving direct inputs (e.g., stormwater discharges, effluent discharges)
- vegetation along the stream bank
- unstable/eroded banks.

Catchment description form

A catchment description allows you to understand the current uses, values and threats to the streams in your catchment.

The form on the NZ Water Citizens website can help you to think broadly about the health of your catchment and will be useful for selecting monitoring locations and interpreting your results. You may want to expand on some sections or leave some sections blank and fill them in later when you have more information. As you fill out the catchment description form, add relevant information to your catchment map. We recommend asking your regional council for help with completing this catchment description and creating maps.

Keep your catchment description along with any other information about your catchment (e.g., maps, council reports, articles or news stories) together in a safe place. As you learn more about your catchment, revisit your catchment description and add to it.

WHERE TO MONITOR

The number and location of sites you choose to monitor depends on your monitoring goals. This section helps you decide how many monitoring sites you need and where to locate them.

The location of your main monitoring site may be already decided if:

- you are interested in a particular site on a particular stream (e.g., a popular swimming site, the stream that runs past your house/farm property/school/park/marae); or
- you want to learn how to monitor and need the nearest accessible stream.

Even so, you may want to add other sites for comparison (e.g. a "reference" or "control" site – see below).

You will need to plan carefully where to locate your monitoring site(s) if:

- you are interested in a whole stream (or a long section) rather than a particular place on it
- you want to investigate the effects of a particular activity or land use
- you want to determine where in a catchment the greatest impact on a stream is occurring
- you want to involve all the landowners in your catchment in learning about/improving stream health; or
- you want to add useful data to council monitoring.

Check your stream is suitable for monitoring

SHMAK has been designed for use in streams that are safely wadeable under all but high flow (flood) conditions. This normally means a maximum of 0.5 m deep, though up to 1 m deep may be fine if the current is very slow. During flood flows wading might not be safe even in quite small streams (though in some places sampling can be done safely from a bank or bridge).

Deciding on the number of monitoring sites

The number of monitoring sites depends on your monitoring goals.

- If you are monitoring for **education and awareness-raising**, to **describe the current state** of a stream compared to a guideline (e.g., to determine if it is safe to swim in), or to **contribute to regional monitoring,** you may only need to monitor a single site.
- To **assess an impact** (determine whether a particular activity, land use or discharge is having an impact) or **evaluate a restoration project**, you need to monitor at least two sites – a "*control*" site upstream and an "*impact*" site downstream of the area where the impact is suspected (see definitions below). If there are no suitable control sites upstream of the suspected impact, a site on a nearby stream of similar size and character, but lacking the impact, may be suitable for a control. A nearby "*reference*" site (see below) could also be useful to see how much your impact site differs from natural condition.
- To assess the effects of your own farm on a stream that flows through it, you will need an "*inflow*" and an "*outflow*" site.
- If you are **investigating an issue of concern**, you might need several sites around the catchment, upstream and downstream of the possible sources of the issue.
- If you are monitoring to **detect trends over time**, we recommend monitoring a "*reference*" site as well as your main site of interest. A reference site provides a benchmark to compare stream health and changes in stream health indicators over time at your main site.

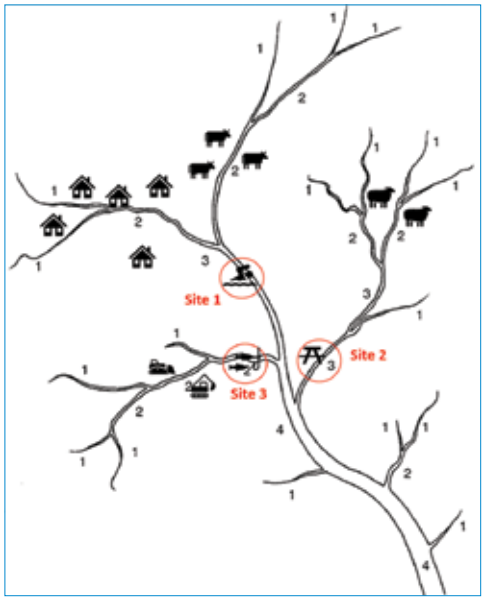
Types of monitoring sites

Impact sites (also called test sites) are sites affected (or suspected to be affected) by a disturbance such as a particular land use or pollution source. The 'disturbance' could also be a positive activity such as riparian restoration.

Control sites are sites that are identical in all respects to the impact site except for the disturbance or activity of interest. They are typically upstream of the impact site or on a very similar stream nearby.

Inflow and outflow sites are located where a stream enters and leaves the area being assessed. They can be used to measure the effects of a single land use (e.g., a farm) on a stream. The inflow site is equivalent to a control site, and the outflow site equivalent to an impact site.

Reference sites are on a similar type of stream to your impact (test) sites but are as natural as possible. Monitoring reference sites allows you to assess how different your impact site is from natural condition, and (over time) to separate natural variation from changes due to human influences.



Case Study: Monitoring to describe the current state of specific stream sites versus monitoring for impact assessment

Example 1. Monitoring to describe the current state.

You are interested in knowing the water quality at popular recreational sites in your catchment (Sites 1-3). Monitoring these sites can provide information on its suitability for different types of recreation (e.g. swimming and boating). You can compare data you collect to relevant water quality guidelines but without control sites or reference sites it will likely be difficult to find the source of any pollution you detect.

Example 2. Monitoring for impact assessment

You want to determine if different disturbances in the catchment are having an impact on your favourite recreational sites. For each impact site, you also have a control site upstream of the impact. Some sites won't have an upstream control and so you will use a similar site on a neighbouring stream. You may also want to target specific impacts, for example urban impacts (Site 7) versus farming impacts (Site 8).

Selecting sites: step by step

Step 1: Review your map

The first step in selecting a site or sites is to review the map(s) of your stream and catchment that you made earlier. Highlight the streams you are most concerned about. The annotated map will help you to decide where to carry out the monitoring. It will also provide a record of your monitoring site locations once they are chosen. At a later stage, after you have carried out your monitoring, the information on the map may help you interpret your results.

Step 2: Mark possible monitoring sites on the map

Mark a rough location on the map for each control, impact, inflow/outflow and/or reference site that you need. Do this for each stream that you are concerned about. Think about how easily you can access the streams at the locations you have marked, and whether they are "wadeable."

Consider monitoring alongside professionals at any professionally-monitored sites nearby. If your data agree closely with theirs at this site, then you can be confident that your monitoring data at all your sites are reliable.

Estimate the time required to visit all your sites. Does the overall time commitment seem feasible if long-term monitoring is an aim? It's better to start with a few sites that you can monitor regularly, rather than over-commit yourself with too many. You can add more after you have gained confidence.

Step 3: Verify your monitoring sites

Next, visit your stream(s) to visually assess their condition, check they are accessible (wadeable, safe to enter, you have landowner permission) and ensure they are suitable for the indicators you would like to monitor. Bring your catchment map so if your first choice of sites aren't suitable you can find others.

Assign a name to each site you have selected, e.g. *"Shady Stream site 1"*. This site name (or a suitable code such as "SS1") will be used for all future monitoring.

WHAT TO MONITOR

Match indicators and methods with your goals

To avoid unnecessary effort, choose indicators that are most relevant to your monitoring goals, the issues or concerns you have about your stream, or the activities that could be causing an impact. Tables 2-1 to 2-3 outline the indicators you might choose to achieve different goals.

Some indicators in SHMAK have two methods – a quick one and an advanced (more detailed and accurate) one. Choose the method that matches your goals and the level of data quality you need to achieve them.

Consider practical factors

Your selection of indicators and methods also depends on practical factors including the equipment you have access to, number of people available, the time you can spend monitoring, your experience and confidence, and the money you have available to purchase equipment and consumables.

Table 2-1. Linking your monitoring goals with possible indicators.

MONITORING GOAL	POSSIBLE INDICATORS
EDUCATION AND AWARENESS	
Some indicators are easier to measure than others, and some have an easier and a more detailed method. Choose indicators and methods that match the skill level of your group. Some indicators take longer to measure than others. Choose ones that fit your timeframe. Aim for indicators that capture people's imaginations or that illustrate a particular teaching point.	Visual clarity, temperature, and conductivity are easy and quick Benthic macroinvertebrates and fish always grab people's interest Nitrate, phosphate, *E. coli* bacteria and rubbish are highly topical Visual clarity, nitrate and *E. coli* tests are very engaging
DESCRIBE THE CURRENT STATE	
Choose indicators that have standards or guidelines to protect ecological, recreational or other values for comparing your data to.	Visual clarity, temperature, dissolved oxygen, *E. coli*, periphyton (attached algae), benthic macroinvertebrates
ASSESS AN IMPACT, INVESTIGATE AN ISSUE, EVALUATE A RESTORATION PROJECT OR DETECT TRENDS	
Choose indicators that match your concerns and the likely effects of different activities or land uses	See Tables 2-2 and 2-3 below
CONTRIBUTE TO REGIONAL MONITORING OR SCIENTIFIC RESEARCH	
SHMAK indicators have been chosen to match those monitored by councils.	Monitor as many indicators as possible. Be aware that nitrate and phosphate may not be accurate at low levels and won't be directly comparable with council lab data.

Impact assessment

If your monitoring goals involve assessing the impact of a particular land use, land management or activity on stream health, then choose indicators that are likely to measure the impacts related to that activity. Table 2-2 shows the types of impact caused by different land uses or activities.

Table 2-2. Examples of different activities and the impact they may have on stream health. Impacts with an asterisk cannot be measured directly using SHMAK, though they will affect stream life indicators.

ACTIVITY	IMPACT ON STREAMS
Agriculture – cropping	Fine sediment, increased nutrients, loss of riparian vegetation, increased water temperature
Agriculture – livestock	Fine sediment, increased nutrients, loss of riparian vegetation, increased water temperature, faecal contamination, bank erosion
Construction	Fine sediment, rubbish, stream habitat alteration, barriers to fish migration
Forestry	Fine sediment, increased nutrients, loss of riparian vegetation, habitat alteration, increased water temperature
Industrial discharges	Toxic contaminants*, increased nutrients, organic compounds**
Urban development	Loss of riparian vegetation, faecal (and other) contamination from stormwater and wastewater inputs, altered stream habitat, barriers to fish migration, rubbish, increased water temperature

*Toxic contaminants include a wide variety of substances that could kill stream plants and animals.
**Organic compounds are substances that use up oxygen as they are eaten by bacteria.

Evaluating restoration

If you are undertaking stream restoration, the goals of your restoration will inform your choice of monitoring indicators. In Table 2-3 we outline different restoration goals, and the indicators relevant to each goal. We recommend reading more about these in ***The Restoration Indicator Toolkit*** (Parkyn et al. 2010), and also seeking professional advice. It may be wise to measure many indicators at the start (e.g. the full SHMAK range), then reduce the number to those you can keep monitoring regularly. In case you find later that some of those first indicators are important, you have some data to act as a "baseline" for comparing to later years.

Table 2-3. Suggested monitoring indicators relevant to different restoration goals. The goals refer to the main value you are hoping to restore.

NATURAL HABITAT	BIODIVERSITY	WATER QUALITY	RECREATION	DOWNSTREAM IMPACTS
Visual clarity	Visual clarity	Visual clarity	Visual clarity	Visual clarity
Temperature	Temperature	Temperature	Temperature	Temperature
Periphyton	Dissolved oxygen	Dissolved oxygen	*E. coli*	Nutrients
Macrophytes	Periphyton	Nutrients	Periphyton	*E. coli*
Streambed composition	Macrophytes	*E. coli*	Rubbish	
Stream habitat	Benthic macroinvertebrates	Periphyton		
Rubbish	Fish	Benthic macroinvertebrates		

WHEN TO MONITOR

How often you visit your site will be determined by your goals and the monitoring indicators you choose. If your goal is to look at a stream's suitabilty for swimming, then weekly monitoring during the bathing season is usually needed (as some indicators used to measure "swimmability", e.g. *E. coli*, can change quickly). If your goal is education, the timing of monitoring visits may be determined mainly by students' timetables (though to compare results from year to year it is best to monitor at the same time each year).

For most other goals, the guidelines below can be used.

ANNUAL	MONTHLY	CONTINUOUS
Benthic macroinvertebrates	Water quality indicators	Streamflow
Macrophytes	Periphyton	Water temperature
Fish	Rubbish	Dissolved oxygen
Stream habitat indicators		Conductivity

Annual indicators - These are indicators that are relatively stable over a year or are most relevant in a particular season. Monitor in the same month every year to ensure you can compare between years. Fish abundances are quite seasonal due to migrations. Macrophytes grow more in summer, and benthic macroinvertebrates are slightly more abundant in summer. Summer is the typical season for monitoring all of these.

Monthly indicators – These are indicators that show much greater variation during a year. Monthly monitoring is needed to smooth out the variations and find average levels. Monitoring at the same time each day is important for indicators that change noticeably over a day (e.g. temperature, conductivity, dissolved oxygen and pH). For periphyton and *E. coli*, you may choose to monitor monthly during the summer period only.

Continuous indicators – These are indicators that can change a lot over a single day. Continuous monitoring requires a "logger" that takes measurements automatically every few minutes (or other interval). Continuous streamflow data from your stream or one nearby may be available from your regional council.

Practical matters

The schedule described above is the one used by regional councils and is recognised as "standard practice" by professionals. So, it is the ideal if you can manage it. But monitoring water quality monthly is time-consuming. If your (or your group's) time is limited, you are free to choose your own schedule. It is better to set a slightly less demanding schedule that you can stick with for the long term rather than burning yourselves out after a few months. One good compromise may be to reduce water quality monitoring from monthly to quarterly. Be aware, though, that it will then take longer to build up enough data to understand variability, to determine average values and to see trends.

On the other hand, monitoring macroinvertebrates once a year may be not frequent enough, as it's easy to forget the different macroinvertebrate types over a year. Quarterly monitoring may help to keep you familiar with the identifications.

High flows and floods

Data collected during floods are particularly useful because floods carry high amounts of fine sediment and other contaminants. However, do not attempt to enter a stream in flood. Instead, take water samples from a bridge (if there is one, using a bucket and rope). If there is no bridge but there is safe access to the stream bank, use a pole sampler. A pole sampler can be made using a telescoping handle, such as a window-washer's pole brush, with the sample bottle taped or wired to it. Indicators usually measured by placing equipment in the stream (e.g. temperature, conductivity, black disc for visual clarity), should be measured on water samples collected in a bucket (provided this is safe). For visual clarity this would require the clarity tube method.

How long to keep monitoring

The longer you can keep monitoring, the more useful your data will be. This is for two reasons. First, all indicators change naturally over time, so you need to monitor several times to work out average values. We recommend at least three years of monitoring to have confidence in your results for **characterising current state** of your stream. Second, it often takes many years (10+) to see changes in stream condition ~~following these changes.~~ Some indicators change faster than others, but all will need at least five years to **detect trends** over time.

If your goal is to **evaluate the success of stream restoration**, we recommend at least two years of pre-restoration monitoring to get a good set of baseline data. For more information, consult **The Restoration Indicator Toolkit** (Parkyn et al. 2010) and your local regional council.

DATA QUALITY

Ultimately you need to ensure that the data you collect is of high enough quality to answer the questions you are asking about your stream. Therefore, the quality you aim for depends on your monitoring goals and who will be seeing or using your data. Generally, if you hope your data will influence (or be used by) others, it will need to be higher quality than if you are using it only for your own information. Table 2-4 is a guide to the minimum data quality needed for different goals and uses. Of course, for any goal, the higher the data quality, the more useful it will be. The highest-quality volunteer data can be defined as being closely comparable to data produced by professionals.

Table 2-4. Minimum data quality needed to achieve different monitoring goals. Use these levels in conjunction with Table 2-5.

GOAL	MAIN INTENDED USE OF DATA	MINIMUM DATA QUALITY LEVEL
Education: introduce stream ecology and monitoring methods, raise awareness	None. Emphasis is on doing rather than on the data itself	Demo
Education: a project involving data analysis	Yourself/your group	Fit for own use
Quick assessment to identify places for further investigation	Depends	Fit for own use
Evaluate a restoration project	Yourself/your group	Fit for own use
Assess your own land management	Yourself/your group	Fit for own use
Describe current state, assess an impact, investigate an issue of concern	Shared with community	Fit for shared use
Contribute to regional monitoring or scientific research	Shared with researchers/ council/public	Fit for shared use
Influence freshwater decision-making	Shared with decision-makers	Fit for shared use

Data quality concepts

Data quality is made up of a number of concepts and the "jargon" can get quite confusing. Here we outline the main concepts relating to data quality. Other terms, less-often used, are in the glossary.

Accuracy: How close the measurement is to the "true" value. The smaller the difference between the measurement and its "true" value, the more accurate the measurement. Accuracy can be determined by measuring a sample that has a known value, such as a standard reference sample from a lab, and comparing the measured value to the known (true) value. However, in the natural environment, the true value is not known, so accuracy cannot be assessed.

Reproducibility: Where accuracy cannot be assessed, the best that can be done is to assess reproducibility. This is where two different people or agencies working independently, get similar results. Reproducibility implies (but does not quite prove) accuracy.

Precision: How similar repeated measurements are when collected by the same person or someone else in your group. Repeated samples or measurements are often called replicates. Like reproducibility, precision does not tell you how accurate a measurement is.

Bias: Where your measurement is consistently higher or lower than the true value.

Quality assurance (QA): The overall plan, including the study design, monitoring protocols, training, quality control, and data management, that promotes data quality. QA begins before you even step in the stream.

Quality control (QC): The activities that are in place to control error while you are conducting your monitoring (e.g., collecting replicates, checking field instruments). These measures ensure the monitoring results are representative of the overall condition of the sample area. QC procedures can be internal (done by project volunteers) or external (done by outside professionals).

Training

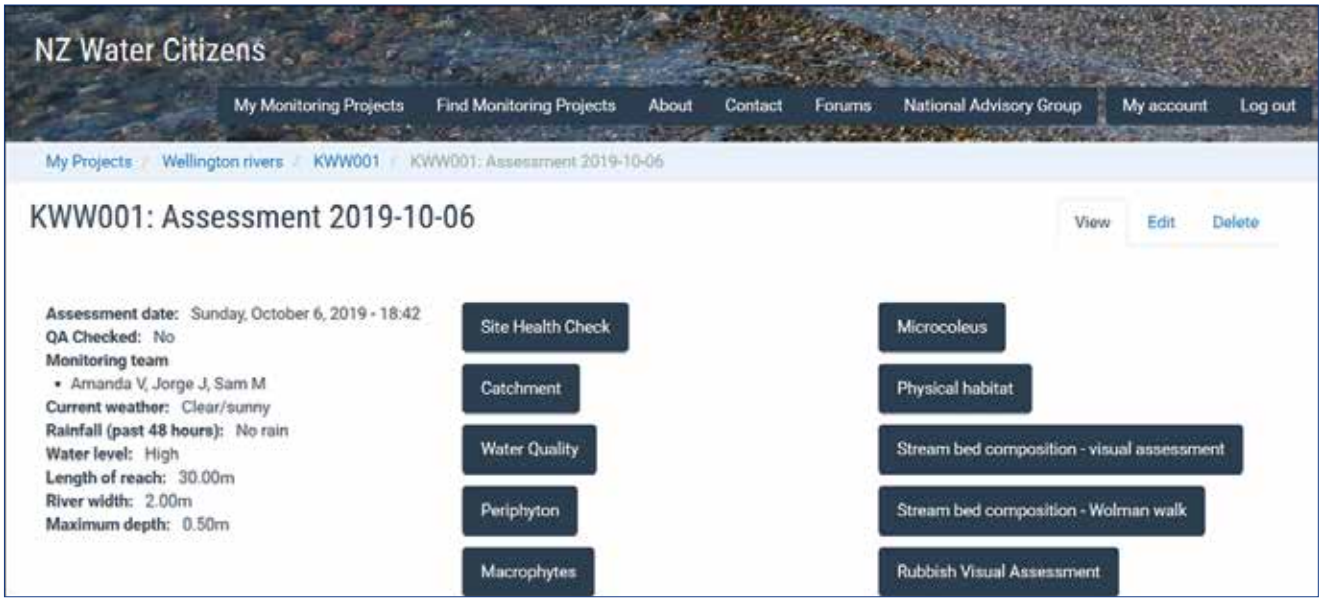
Hands-on training with a qualified instructor is key to collecting high quality data. A SHMAK training course will provide you with the necessary skills to perform all the monitoring methods outlined in this manual in a scientifically robust way. Some aspects, for example, *E. coli* methods or identification of benthic macroinvertebrates, require extended training due to their complexity. **Check the NZ Water Citizens website** to learn about training opportunities in your area. For those who can't access face-to-face training, the website also has training videos and links to other training resources. For ongoing learning, there are facts sheets, articles, web links and forums for discussions with experts and other volunteers.

Nominate a training coordinator in your group who will:

- keep up to date with training opportunities
- ensure that at least one trained person is on every sampling trip, and
- keep documentation of everyone's training.

Quality Assurance and Quality Control

Once you have decided on the level of data quality you need (relative to your monitoring goals and the uses of your data, Table 2-4 above), decide which QA/QC steps you need to reach that data quality. These steps are outlined in Table 2-5.



Internal Quality Control Procedures

Field replicates: Field replicates are two or more measurements or samples collected and tested from the same site by the same person or by two people in your group. By collecting field replicates, you can assess your performance and the precision (repeatability) of your results. For some field measurements, such as visual clarity, the method always involves two measurements. If the measurements from replicates are more than about 10% different, it could indicate differences in the way each individual is measuring. For *E. coli* and other laboratory measurements, it may be too costly to collect replicate samples every time. If so, when developing a monitoring plan, decide how often you will collect replicate samples (e.g., every 5th or 10th sampling event).

Voucher specimens: Vouchers can be physical specimens or photographs. For benthic macroinvertebrate monitoring, you can preserve a set of at least one good specimen (preferably 3 – 5) of each taxon found at a site. Your identifications can then be confirmed by an expert. Photographs, provided they are of high quality (in focus, high resolution and showing the relevant body parts), are useful if you want to release the animals back into the stream.

Blanks: A blank is a sample (usually of pure water) that you would expect to give a "zero" measurement. It is a way of checking for contamination during sample handling and analysis, and that your equipment is correctly "zeroed". A field blank is put into a sample bottle in the field, and is a check on your field and lab methods. A lab blank is put into a sample bottle in the lab and checks only your lab methods. In SHMAK, lab blanks are recommended when testing for *E. coli*. A sample bottle is filled with tap, bottled or distilled water and then analysed for *E. coli* using the same method as for a stream water sample.

Standards: A standard is a dissolved chemical solution made up in a lab to a known concentration. You can check the accuracy of a meter (such as your conductivity meter) by measuring a standard with the meter. If the meter reads too high or low, you can adjust it (calibrate it) to read the value of the standard. A chemical test such as for nitrate or phosphate can be checked in the same way. If these are not giving accurate readings, check the expiry date of the chemical test packets.

Data management: Data management represents the systems you have in place to ensure proper sampling information (date/time, site name, flow conditions, etc.) is recorded, data entry and transcription errors are minimised, missing data are noted, and data and documents are stored safely.

Choose someone in your group to check in the field that data sheets are complete and data look correct, and someone to upload data to a computer and check that no errors have been made in uploading. The NZ Water Citizens website www.nzwatercitizens.co.nz can store information about your site, details about each monitoring trip and the data collected on each trip. Other information, such as your monitoring plan, background information and maps, should be stored in a safe place (e.g., in a filing cabinet or on a shared computer drive). Make sure more than one person knows where the information is kept.

External Quality Control Procedures

External quality assurance and quality control (QA/QC) will pick up any issues with how you take measurements that could lead to errors in your data.

Field replicates: Field replicates are collected by sampling side-by-side with a professional such as regional council monitoring staff. If your results agree, this shows you can collect "reproducible" data, giving you confidence in your methods.

Data review: Asking an outside partner or agency to review your data is another effective quality control measure. This is especially important if you would like the agency to use your data. An external review can help identify gaps in your quality control efforts. It also ensures that your data collection and reporting activities make sense to others.

Auditing: A formal audit by a professional agency is probably the most thorough external QA/QC procedure. It would typically include field replicates and data review (described above) as well as checking that all field and lab methods are being followed correctly.

Table 2-5. Recommended QA/QC procedures for three levels of data quality.

QA/QC PROCEDURE	DATA QUALITY LEVEL		
	DEMO	FIT FOR OWN USE	FIT FOR SHARED USE
MONITORING PLAN			
Monitoring plan developed and recorded		Template filled in	Reviewed by professional
TRAINING AND AUDITING			
Training	Follow instructor's directions	Read manual, watch videos	Training course completed
Audit by trainer or professional			Once every 2 years
DATA COLLECTION			
Standard equipment used	For every monitoring event	For every monitoring event	For every monitoring event
Equipment is working, chemicals not expired [1]	For every monitoring event	Before every monitoring event	Before every monitoring event
Equipment calibrated or validated		Conductivity meter calibrated	Also nutrient tests validated (10%)
Containers and equipment cleaned/ sterilised [2]		For every monitoring event	For every monitoring event
Method level	SHMAK level 1	SHMAK level 1	SHMAK level 2
Record date, location, weather and flow conditions	For every monitoring event	For every monitoring event	For every monitoring event
Visual assessments done in pairs [3]		Whenever possible	For every monitoring event
Tests/observations repeated [4]		Clarity	Also 10% of water quality samples
Tests/observations repeated by a professional [5]		If possible	All indicators. Once per 2 years.
Samples stored and transported correctly	For every monitoring event	For every monitoring event	For every monitoring event
Lab blanks used for *E. coli*		On 10% of samples	On 10% of samples
DATA MANAGEMENT			
Data checked in field – correct and complete	For every monitoring event	For every monitoring event	For every monitoring event
Data stored securely		For every monitoring event	For every monitoring event
Data transfer to spreadsheet/ website checked		For every monitoring event	For every monitoring event
Data verified by expert			Checked regularly

[1] relevant equipment: clarity tube and black disc viewer (check for scratches on viewing window), temperature/conductivity meter (check correctly working), nitrate kit, phosphate checker, *E. coli* plates (check correct storage and expiry of chemicals), kick net (check for holes).

[2] sample containers for conductivity, nitrate and phosphate require cleaning. Containers and equipment for *E. coli* require cleaning and sterilising.

[3] two people discuss their estimates and agree on the value to be recorded. Relevant to visual assessments of cover (periphyton, macrophytes, stream bed composition), identification of bugs and fish, rubbish visual reach assessment.

[4] tests for "repeatability" or "precision".

[5] you and a professional monitor side by side. Either you go to a professional monitoring site, or the professional comes to your site. Tests for "reproducibility".

GIVING EVERYONE A ROLE

Running a successful stream monitoring project involves many roles and responsibilities. It is important that everyone in your monitoring group knows what tasks they are responsible for and the effort is shared among members. Think about the different tasks that need to be accomplished and agree which members of your group will take on which roles, according to their interests and skills. Examples of possible monitoring roles include:

- **Project Coordinator:** The overall leader. Keeps track of the monitoring plan, reminds volunteers of monitoring dates on the schedule, convenes group meetings and training events.
- **Health and Safety Coordinator:** Oversees the development, putting in place and periodic review of a Health and Safety Plan that ensures all group members know how to stay safe when monitoring.
- **Training Coordinator:** Understands the monitoring methods and regularly upskills and attends refresher courses. Although we recommend that everyone attend a training course, your group may not have the resources to send everyone. The training coordinator could become a trainer by taking a "train-the-trainer" course when available and periodically re-watching the SHMAK training videos.
- **Equipment Coordinator:** Stores monitoring equipment, checks expiry of reagents, ensure field equipment is working, orders new consumables and replaces damaged or broken items.
- **Data Management Coordinator:** Collects and stores data sheets, enters data into database, checks for missing data, conducts data analysis and reports back to the wider group. Flags any issues to be raised with the regional council or other support agency.
- **Data Entry Assistant:** Assists with data entry, particularly checking that the all the data from datasheets have been entered into the database correctly.

Multiple roles might be performed by the same person.

DEVELOPING YOUR MONITORING PLAN

A monitoring plan gives an overview of your project, including all the decisions you made in designing your project. The monitoring plan will ensure that everyone involved in the monitoring project understands why the data are being collected and how it will be used. You should nominate a project coordinator who will lead the development of the plan and review the plan, say annually, to ensure it continues to meet the needs of the project. The value of your data is directly related to how much effort you apply in developing your monitoring plan.

A monitoring plan template can be found on the NZ Water Citizens website.

SOURCES OF INFORMATION

Land, Air, Water Aotearoa (LAWA)

LAWA presents information on freshwater and beach water quality, freshwater quantity, air quality and land cover. You can search for river monitoring sites in your region. Not all sites monitored by regional or unitary councils can be found on LAWA but it is a good starting place. LAWA also provides information on land cover (vegetation) from the New Zealand Land Cover Database (LCDB)

Ministry for the Environment (MfE)

You can browse and download information on a wide variety of information sources about your region. This includes a list of vulnerable catchments, climate data (sunshine hours, average rainfall, etc.), location of aquifers, and land use maps. MfE also publishes a report every three years on the state of New Zealand's fresh waters as part of their Environmental Reporting Series.

NZ Landcare Trust

NZ Landcare Trust works with landowners from across the country and support catchment groups in restoring and monitoring streams. NZ Landcare Trust may have been involved in a project on your stream or in your catchment. Their regional webpages provide more detailed information on projects and the contact details of the NZ Landcare Trust coordinator in your region.

NIWA

New Zealand Freshwater Fish Database (NZFFD)

The New Zealand Freshwater Fish Database contains over 34,000 freshwater fish observations. Data stored include the location of sample sites, the fish species present, as well as information on their abundance, size, sampling methods and a physical description of each site. You can search for fish species found in your catchment.

National Climate Database

The climate database receives data from over 600 climate stations across the country. It includes data on rainfall, temperature, sunshine, frost, wind and rain. Search the database to determine your closest climate station.

National River Information

NIWA monitors river flows and water quality at a number of sites across New Zealand. Use NIWA's Hydro Web Portal to explore all data locations using a map. Select the sites in your catchment to view and export available data.

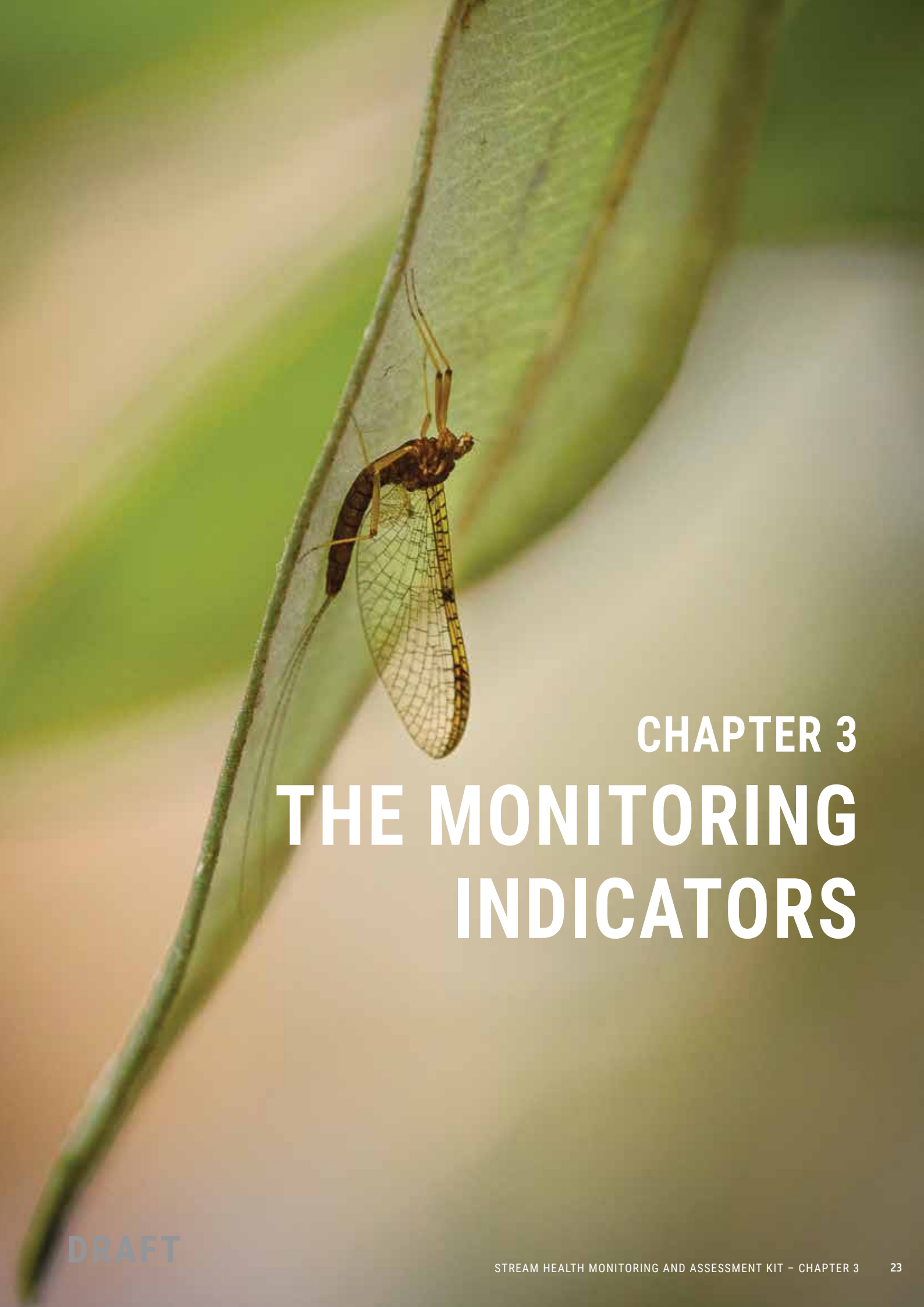
NIWA has also produced NZRiverMaps, an interactive web-based tool for exploring national scale estimates of various river-related properties, including water quality, hydrology, bed sediment size, invertebrates, fish presence, bed sediment cover and water allocation.

Our Environment – Manaaki Whenua Landcare Research

Our Environment provides access to Manaaki Whenua Landcare Research's environmental data. You can use online, interactive maps to learn about geology, soils, vegetation, land use etc. in your area, and create custom maps to help you select monitoring sites.

NZ Water Citizens

The NZ Water Citizens website stores data that have been entered by other volunteer monitoring groups. You can view, graph and download data from other groups in your area.



CHAPTER 3
THE MONITORING INDICATORS

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INTRODUCTION

In this chapter, three categories of monitoring indicators are presented; water quality, stream life (aquatic plants and animals) and stream habitat.

Before you decide what to monitor, it is important to read through this section and understand what each indicator can tell you about the health of your stream.

Stream life and water quality differ in some important ways, and therefore tell us slightly different things about the health of a stream.

Water quality samples

- reflect conditions throughout the whole catchment: influenced by all the waters entering from different sources upstream
- a "snapshot" in time: may not capture a once-off pollutant spill, or extreme values that occur at a different time of day to your monitoring visits
- indicate what may be passed on to downstream waters (rivers, lakes, estuaries, the sea)

Stream life

- respond mainly to the habitat and water quality immediately around them
- can reflect conditions over weeks or months: influenced by conditions that occur when you are not there
- give a picture of overall stream health but it can be hard to know what is affecting them most (because they respond to a wide range of water quality and habitat conditions)

Monitoring water quality, stream life and stream habitat quality together gives the most complete picture of overall stream health, and each indicator helps you to understand or interpret the others. For a few indicators we have two levels (methods) of assessment.

VISUAL CLARITY

Why is it important?

Visual clarity is a measure of underwater visibility in streams. It reflects the concentrations of fine sediment, algae, and other particles suspended in the water. Reduced visual clarity can harm aquatic animals and river birds who rely on sight to find prey and avoid predators, and swimmers who may not see underwater hazards. Reduced clarity also means less light getting through the water to the stream bed where plants need it for photosynthesis. The fine sediment that causes low visual clarity may clog the gills of fish and benthic macroinvertebrates.

How is it measured and reported?

Visual clarity is measured using a clarity tube or black disc, which both measure the distance through water that a human eye can see a black object. The results are reported in metres. The measurement value to be reported is the average of two measurements, the distance at which a black disc disappears from sight and the distance at which it reappears. Most streams are best assessed by viewing a black disc in the stream through an underwater periscope. This equipment works well down to about 0.1 m (10 cm) visibility. If your stream is very turbid (<0.5 m visibility), the clarity tube is preferred because it is more precise and safer (no wading required). Visual clarity is strongly influenced by streamflow, and so is very changeable and should be measured on every visit (e.g. monthly).

What does it tell us?

Low visual clarity (cloudy water) usually tells us that fine sediment such as clay and silt particles are getting into the stream. Human activities in the stream catchment (such as earthworks or livestock grazing) can greatly increase the amount of fine sediment that enters the stream. Low visual clarity is often accompanied by faecal contamination, nutrients and other contaminants, so it may also indicate that levels of these contaminants are high.

WATER TEMPERATURE

Why is it important?

Aquatic animals struggle to live in warm water. In New Zealand, benthic macroinvertebrates cannot live in water warmer than about 22 °C, and few aquatic animals can survive above 30 °C. This is because they are used to living in well-shaded forested streams, and they are not used to the high afternoon temperatures that occur where that tree cover is gone. So, small streams without much shading usually have only a few species of aquatic animals.

Water temperature also affects many of the physical and chemical characteristics in streams. Warmer water holds less oxygen than colder water and increases the sensitivity of aquatic animals to toxins and diseases (two reasons why aquatic animals die in warm water), and increases the rate of chemical reactions and biological processes such as photosynthesis.

How is it measured and reported?

Single measurements of temperature are made with a thermometer. We recommend measuring water temperature at the same time of day on each monitoring visit, and always recording the time that you measure. This is because water temperature fluctuates over the day, reaching a minimum near dawn and maximum in the afternoon. A single temperature measurement may not be very informative. A series of measurements over time will show how stream water temperature changes with weather and season, and allow you to compare your stream with others.

However, even a series of measurements may not show the extreme temperatures that cause stress on aquatic life. For that, you need to measure at mid-afternoon on the hottest days - days with clear sun during late summer when stream flows tend to be lowest. Temperature loggers are often used to capture the full range of water temperatures that occur over a day and during the hottest days. Temperature loggers are set to measure temperature at regular intervals (usually every 15 to 30 minutes) for a set period of time (usually weeks or months).

What does it tell us?

Many human activities can change water temperatures, including:

- discharging warm water from thermal power stations
- releasing water from dams (releasing bottom water can reduce downstream temperatures, releasing surface water can increase them)
- removing shade trees and shrubs from riparian areas
- changing water levels by abstraction or diversion of water for irrigation, and
- connecting paved surfaces to streams via stormwater pipes.

CONDUCTIVITY

Why is it important?

Conductivity is a measure of how well the water can conduct an electrical current. Conductivity increases with increasing salt content (salts such as chloride, bicarbonate, sodium, calcium,) and increasing temperature.

Each stream tends to have a relatively consistent range of conductivity. Once you know this range, you can use it as a baseline to compare with each conductivity measurement. A large increase in conductivity (if the water level has not changed much) might indicate that a discharge or some other source of pollution has entered a stream.

The dissolved salts measured by conductivity usually do not have a direct effect on stream life until they reach levels found in brackish water or seawater (greater than about 5,000 µS/cm).

How is it measured and reported?

Conductivity is measured using a conductivity meter and is reported in microSiemens per centimetre (µS/cm) (the opposite of electrical resistance). Because conductivity is higher at warmer temperatures, the displayed value is adjusted to what conductivity would be at 25 °C. Conductivity measurements are most useful when collected many times over a long period. Recording the typical range of values at different times of year and during different stream flow conditions allows you to notice any unusual changes outside of this range.

What does it tell us?

Conductivity is often used in water quality studies as a quick indication of the level of salt content (including nutrient "salts") in the water. One particular use is detecting intrusion of seawater or geothermal water, since both of these have much higher conductivity than stream water. If you are looking for inanga spawning areas, this can help you locate the end of the "saltwater wedge" where inanga usually spawn.

Conductivity can also tell us something about where the stream water has come from. High-conductivity streams typically have more input of groundwater, with longer underground flow paths, than low-conductivity streams.

In general, however, conductivity is hard to interpret on its own, and is normally used to support other information. For example, high conductivity may be a factor explaining high growth of periphyton (algae) in a stream, but you need to confirm that all other conditions (e.g. light, temperature, streamflow) are suitable for growth and have recorded high periphyton cover there.

NITRATE

Why is it important?

Nitrogen is an important nutrient supporting plant growth. Nitrogen in soil and water is cycled through several different forms, including ammonia/ammonium (NH_3/NH_4+), nitrate (NO_3-), nitrite (NO_2^-) and organic nitrogen. Nitrate dissolves easily in water and can easily pass through soils, particularly during heavy rainfall. Once in a stream it can be readily used by plants and algae for growth and even moderate concentrations can cause plants and algae to grow to nuisance levels. However, the effects depend on various other factors, such as phosphate (the other important growth nutrient), light, flood frequency, etc. High nitrate concentrations can also be toxic to aquatic animals, and to human health. High nitrate concentrations in drinking water have been linked to a blood disorder in infants called blue baby syndrome.

How is it measured and reported?

Nitrate concentration is usually measured using "colorimetric" methods, where chemicals added to a water sample react with the nitrate to form a colour. The intensity of the colour shows the concentration of nitrate. Professional laboratories measure the colour intensity (actually, the amount of light absorbed by the sample) using an instrument called a photometer or colorimeter. In SHMAK you read the colour intensity by eye using a colour comparator, so the SHMAK method unfortunately is less accurate than professional methods.

Nitrate concentrations are usually reported in mg/L, which is the same as g/m^3 or ppm (parts per million). Normally it is "mg/L nitrate-nitrogen" (or "mg/L NO_3-N"), but sometimes you might see "mg/L nitrate" instead. The difference is that mg/L nitrate-nitrogen measures only the nitrogen atom whereas mg/L nitrate measures the whole nitrate molecule, which is heavier. 1 mg/L nitrate-nitrogen is the same as 4.42 mg/L nitrate.

What does it tell us?

In agricultural catchments, high nitrate concentrations in streams typically result from nitrogen fertilisers and livestock that excrete large amounts of nitrogen in their urine. Near homes and residential areas, high nitrate can also indicate overflows or leakage from sewage pipes or septic tanks, or or it can be found in stormwater inputs. Apart from toxic effects, the main concern with high nitrate in streams is its effect on stimulating periphyton (algae) or macrophyte (aquatic plant) growth. But because nitrogen can cycle between different forms, it is hard to predict the effects of nitrate without also knowing how much nitrogen is in the water in other forms (e.g. ammonium or organic forms). High nitrogen suggests that nuisance growths of periphyton or macrophytes might occur, but other factors, such as low light, low phosphorus, low temperature and/or frequent floods, may prevent nuisance growths. To get a complete picture of nitrogen in your stream, we recommend sending a water sample to a professional lab for ammonium, nitrate, nitrite and total nitrogen analysis.

PHOSPHATE

Why is it important?

Phosphorus is an essential growth nutrient for plants. In water, phosphorus occurs as phosphate (PO_4^{3-}) which has both organic forms (derived from plants and animals) and inorganic forms (derived from rock). Most phosphate enters streams attached to soil particles, some of which settle on streambeds. While it is attached to this sediment, the phosphate is not immediately available as a nutrient for plants and algae. However, over time and in the right conditions it can be released from the sediment as dissolved phosphate, which can be taken up by growing algae and aquatic plants. Even at very low concentrations phosphate can stimulate plant growth.

How is it measured and reported?

Like nitrate, phosphate concentration is normally measured using a colorimetric method. When a chemical is added to the water sample, it reacts with phosphate to form a blue colour and the intensity of this colour is measured using a photometer or colorimeter. Like a professional lab, the SHMAK kit uses a colorimeter (called a phosphate checker), but it is less accurate than a professional colorimeter, especially at low concentrations.

Phosphate concentration is normally reported in mg/L, which is the same as g/m³ or ppm (parts per million). Because phosphate concentrations in streams are often very low, you often see ppb (parts per billion) too. Parts per billion is 1000 times greater, i.e. 1 ppm = 1000 ppb.

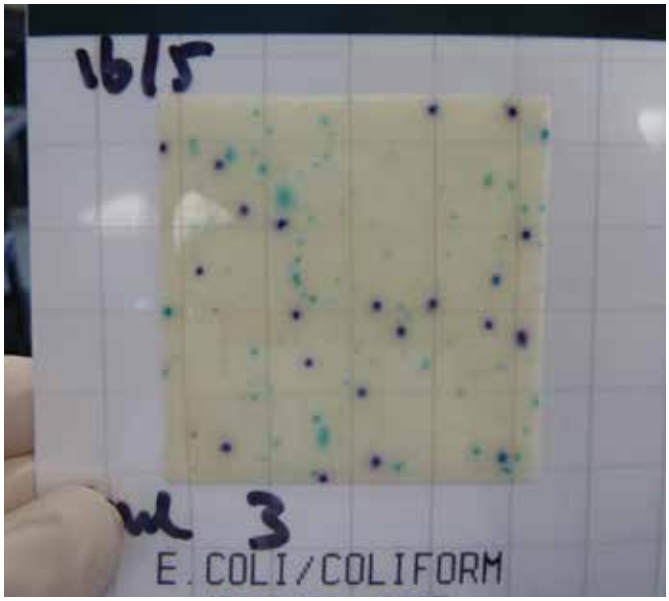
The Hanna Instruments phosphate checker in SHMAK shows phosphate concentration as mg/L phosphate, but most professionals report it as mg/L phosphate-P. The difference is that mg/L phosphate-P measures only the phosphorus atom whereas mg/L phosphate measures the whole phosphorus molecule, which is heavier. Multiplying your colorimeter results by 0.326 will convert them from phosphate to phosphate-P, so you can compare them directly with most professional lab results. 1 mg/L phosphate is the same as 0.326 mg/L phosphate-P.

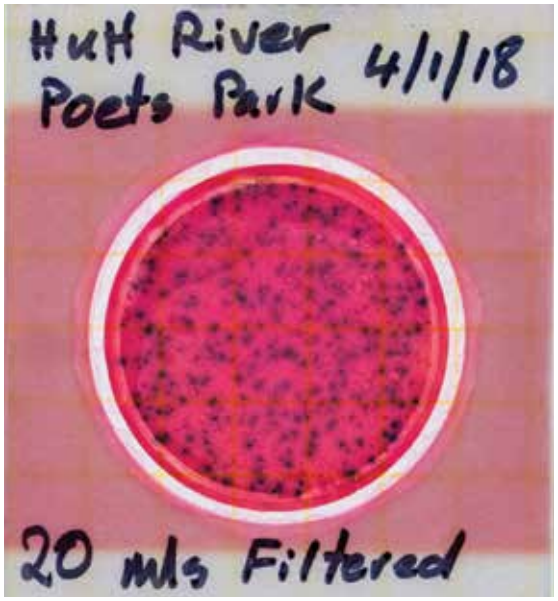
Measuring dissolved phosphate is challenging because in most New Zealand streams it is present in very low concentrations (< 0.01 mg/L).

What does it tell us?

High phosphate concentrations can indicate high levels of soil erosion and/or use of phosphate fertiliser in the catchment. In urban environments high phosphate concentrations can result from wastewater and stormwater inputs, runoff from commercial cleaning or from phosphate fertilisation of gardens and playing fields.

The main concern with phosphate in fresh water is its effect on stimulating algal or aquatic plant growth. However, it is hard to predict these effects from the amount of dissolved phosphate in a water sample, as sediment-bound phosphate (which is difficult for volunteers to measure) may also contribute to plant growth. Even if all the phosphorus is measured, it still may not be easy to predict effects on aquatic plant growth for the same reasons as described for nitrate. To get a more complete picture of phosphate in your stream, we recommend sending a water sample to a professional lab to analyse for phosphate (typically referred to as dissolved reactive phosphorus) and total phosphorus.





E. coli BACTERIA

Why are they important?

Faecal microorganisms come from humans and warm-blooded animals such as farm animals, pets, birds and pest mammals. They can enter streams in a variety of ways including discharge or leakage of sewage, animals defecating in the water, or runoff from grazed pastures. Drinking, swimming or eating certain foods (e.g., shellfish) from water contaminated with faecal matter can make you sick from taking in human faecal pathogens such as *Campylobacter* or *Cryptosporidium*. The bacteria *Escherichia coli* (*E. coli*), found in the intestines of warm-blooded animals, is used as an indicator for faecal contamination. The common strains of *E. coli* are not harmful themselves, but indicate that harmful pathogens may be present.

How are they measured and reported?

E. coli in water is reported as "colony forming units" (cfu) in 100 mL of sample water. Each colony on a growth plate represents a single *E. coli* cell, or a clump of cells, that has multiplied many times and become visible to the human eye. *E. coli* can be grown and counted on a Select *E. coli* Count Plate (SEC plate). The colour of the colonies depends on what brand of growth plate you use. The growth media on these plates contains a combination of nutrients and colour-producing chemicals that make *E. coli* colonies appear purple-blue (MCM media) or blue (Petrifilm™). Coliform bacteria that are not *E. coli* appear green (MCM media) or pink/magenta (Petrifilm™).

What do they tell us?

High concentrations of *E. coli* bacteria suggest that pathogenic microorganisms might also be present in your stream and swimming or water sports may be a health risk. *E. coli*, together with rubbish, periphyton and visual clarity, measures the overall 'swimmability' of fresh water.

Sources of faecal contamination could be humans (e.g. leaking septic tanks or sewers), livestock, waterfowl or, in urban areas, pet waste. The risk of human pathogens differs between these different sources. For example, the risk from cattle faeces is roughly similar to human sewage, while the risk from bird faeces is likely to be lower. Further investigation will be needed to work out the most likely source of the *E. coli* you measured, and the health risk it represents. Your local regional council or organisations like ESR (the Institute of Environmental Science and Research) may be able to help with this.

PERIPHYTON

Why is it important?

Periphyton refers to communities of algae and cyanobacteria attached to the sediment surface or to aquatic plants. Periphyton varies from thin slippery films to thick mats or long filaments in many shades of green and brown.

Periphyton plays an important role in stream ecosystems by providing a food source for macroinvertebrates. But too much periphyton causes problems. Thick growths look ugly, spoil recreational activities such as swimming and fishing, and can clog water intakes and filters. They can also smother habitat for macroinvertebrates and strip oxygen from the water at night, which harms macroinvertebrates and fish. One type of periphyton (a cyanobacteria or "toxic algae" called *Microcoleus*) can taint drinking water with a musty odour and even produce toxins that have killed dogs.

How is it measured and reported?

In SHMAK, you can choose between two methods for estimating periphyton coverage, depending on the time available and your equipment. The results are reported as the percentage of the stream bed covered by periphyton in a certain area.

The stone method (SHMAK Level 1)

Examine 10–20 stones, one at a time

- Doesn't require equipment
- Quick

The viewer method (SHMAK Level 2)

View 10–20 areas of stream bed

- Covers a larger area
- Provides a more accurate estimate of periphyton cover across your whole site

Because periphyton growth changes a lot over time, it is best to monitor it every time you visit the site (ideally monthly) so you can calculate average growth. Monitoring periphyton at the same time as water quality is useful because periphyton responds to water quality (especially nutrients and temperature). The most important information about periphyton is its maximum growth: how much periphyton accumulates when conditions are ideal (e.g., stable flows, plenty of sunshine, and warm temperatures – conditions that are most likely to occur in late summer).

What does it tell us?

Most small New Zealand streams have little obvious periphyton, because they are naturally shaded and the periphyton is quickly eaten by benthic macroinvertebrates. Blooms of nuisance periphyton are usually a symptom of a system stressed by factors such as nutrient enrichment, high light (from removal of bankside vegetation) and high temperatures (that increase algal growth rates and stress some invertebrate grazers). If enough light is reaching the stream bed, nutrient levels tend to be the main factor limiting periphyton growth. This means that an increase in nutrients can cause periphyton growth to reach nuisance levels (provided floods are not frequent and the stream has a stony bed on which periphyton can easily grow).

MACROPHYTES

Why are they important?

Macrophytes are large aquatic plants, often (but not always) with leaves and roots. In muddy or sandy-bottom streams they are normally the main type of aquatic plant (whereas periphyton is the main plant type in stony-bottom streams). Macrophytes produce oxygen, provide refuge for fish and substrate for benthic macroinvertebrates and contribute to nutrient cycling. However, too much macrophyte growth can impact on human values by affecting swimming or fishing, causing flooding, clogging water intakes, depleting dissolved oxygen levels, smothering the stream bed and causing fine sediment to settle on the stream bed. Some macrophytes, such as oxygen weed, are classed as invasive (or noxious) weeds because they quickly invade new areas and form large dense beds that choke waterways and exclude other plant and animal species.

How are they measured and reported?

There are two macrophyte indices:

- The "macrophyte water surface cover index" estimates the amount of water surface area occupied by macrophytes
- The "macrophyte clogginess index" estimates the percentage of the water volume that is occupied by macrophytes.

Macrophytes are reported as the average cover of macrophytes at your site (as a percentage).

Since macrophytes require some soft sediment (sand or mud) they are typically assessed in soft-bottom streams only. However, some species can be rooted in the stream bank and grow across the water surface in smaller hard-bottom streams.

Macrophytes can be monitored once per year in summer, when their growth is at its peak. But monitoring also in spring and autumn, provides more information, which can be helpful.

What do they tell us?

The amount of macrophyte cover in a stream tells us how suitable the conditions for growth are. These conditions include the amount of light, flood frequency, flow velocity, and nutrient concentrations in the water and sediments. Nuisance growths of aquatic macrophytes are generally most common in unshaded, nutrient-rich lowland streams. Nutrient runoff from the land, as well as inputs from sewage and stormwater discharges, can promote macrophyte growth. Increasing stream lighting to >30% of full sun, (e.g., by removing riparian vegetation), will typically result in nuisance growths. Conversely, restoring riparian forest can limit growths of pest species while enabling shade-tolerant native species to survive.

The two macrophyte indices indicate impact on stream flow, recreational use and ecological health. High macrophyte cover can harm aquatic animals by reducing dissolved oxygen during the night when they respire; measuring minimum dissolved oxygen (typically near dawn) can help determine if this is occurring.

BENTHIC MACROINVERTEBRATES

Why are they important?

Benthic macroinvertebrates are animals that live at the bottom (benthic) of streams and lakes, are large enough to be seen with the naked eye (macro) and lack a backbone (invertebrate). We often call them stream bugs for short. In fresh waters, they consist of the immature stages of many insects such flies, mayflies, caddisflies, stoneflies, beetles and damselflies. As well as insects, there are also crustaceans (e.g. kōura/crayfish and shrimps), snails, worms and leeches. Stream bugs are a key part of stream food webs, feeding on periphyton, macrophytes, dead wood or each other. The aquatic larvae are an important food source for fish and the winged adults are often eaten by birds.

How are they measured and reported?

In the SHMAK, macroinvertebrates are sampled using a sieve or a kick-net as outlined below.

	SHMAK level 1	SHMAK level 2	SHMAK level 2
Stream type	Stony-bottom	Stony-bottom	Sandy/muddy-bottom
Equipment	Sieve	Kick-net	Kick-net
Method	Collect individual stones	Disturb the stream bed to dislodge macroinvertebrates	Jab vegetation, brush logs and wood
Advantages	Quicker than kick-net	More accurate	More accurate
Disadvantages	Misses some species	Time consuming	

After collecting, the different bug types are identified. Each type has a "tolerance score" from 1 to 10 that indicates how sensitive it is to water pollution or habitat degradation (10 means only found in healthy streams, 1 means often found in poor conditions). The different scores can be combined and reported in one or more macroinvertebrate health indices. One index commonly reported by professionals in the Macroinvertebrate Community Index (MCI).

What do they tell us?

Because the tolerance of each macroinvertebrate type to environmental impacts is well known, the particular variety of bugs present in a stream can tell you about environmental conditions there. However, it might not tell you which environmental factor is causing some bugs to be missing. Undertaking a habitat assessment and water quality measurements at the same time can help you work this out. Stream bugs live in streams for months or years so they reflect a range of habitat and water quality conditions over a long period of time, whereas water quality measurements measure only one type of impact at one point in time.

FISH

Why are they important?

Native fish are an important part of New Zealand's freshwater biodiversity. New Zealand has approximately 53 native species of freshwater fish (although the exact number of species is under review), most of which are endemic (found only in NZ). Most species are declining in numbers across the country and some are threatened with extinction.

Fish play an important role as top predators in healthy stream ecosystems. The species present and their abundance affects abundances of stream bugs and some ecosystem processes.

How are they measured and reported?

The three main sampling methods for estimating fish numbers in a stream are backpack electrofishing (pictured top left), trapping (with fyke nets or Gee minnow traps) and spotlighting. Spotlighting is the method recommended in the SHMAK because it requires the least specialised equipment.

Spotlighting (recommended in SHMAK):

Spotlighting is the method recommended in the SHMAK because it requires the least specialised equipment. Spotlighting involves shining a powerful spotlight into the stream reach at night and recording what species are seen. It is suitable for wadeable streams because many freshwater fish species are benthic (bottom-dwelling) and nocturnal (more active at night).

Trapping:

Trapping involves setting a fyke net or Gee minnow trap along the stream reach, leaving it out overnight and retrieving it the next morning. This method is a viable alternative option if working with a council or community organisation that can purchase the equipment.

Electrofishing:

Electro-fishing, which uses an electric current to temporarily stun fish so that they can be assessed, must be carried out by a trained professional.

Whichever method is used, the types and numbers of each species caught are usually recorded in the New Zealand Freshwater Fish Database. Fish surveys are best undertaken during the summer (December to March) as fish generally become less active when temperatures are low. The optimal time for sampling is when flows are stable and at (or close to) base flow. Sampling within 5 days after a flood is usually avoided.

Fish data are usually reported as simply a list of fish present (with their abundances and size range if measured). There is an index called the Fish Index of Biotic Integrity, which increases with more native fish species present and decreases with more pest fish present.

What do they tell us?

The range of native fish present can tell us about the habitat and water quality, both at your site and between your site and the sea. Also, because about a third of native species spend some part of their lives at sea, they need uninterrupted passage between the sea and their freshwater habitats to complete their life cycle. Therefore, species may be absent if there is:

- habitat loss (including loss of water, riparian vegetation, large instream features like boulders and wood, or infilling of the gaps between streambed cobble by fine sediment)
- a barrier to migration (e.g., culverts and dams)
- low dissolved oxygen levels
- high temperatures
- low food resources (benthic macroinvertebrates) or
- predatory introduced fish present.

To know what fish species are absent you will need to know what species should be there. Sampling a nearby reference site or checking the NZ Freshwater Fish Database (**https://nzffdms.niwa.co.nz/**) could help, but best would be to discuss with a fish expert, such as at your regional council.

CURRENT VELOCITY

Why is it important?

Current velocity is the speed at which the water moves in the stream, and is a key aspect of aquatic habitat. A fast current helps plants to take up nutrients and animals to take up dissolved oxygen from the water. And it brings more food to aquatic animals than a slow current. So fast-moving streams often contain a higher diversity of macroinvertebrates and fish than sluggish streams.

However, water velocity also exerts a direct force on plants and animals, so a very fast current can stop periphyton and macrophytes from reaching high abundances. High velocities during floods can flush out fish, dislodge stream macroinvertebrates and uproot macrophytes. Periphyton may be scoured off rocks during high velocities by the water itself, by abrasion of moving sediment (sand blasting) or by the rocks being rolled over.

How is it measured and reported?

Surface velocity can be measured from the rate that a floating object (e.g., an orange) moves along a measured length or section of stream.. It is best to measure in a run section where flow is fairly uniform and there are not many obstructions.

This method gives only a rough measure of average velocity because water moves at different speeds in different places: it is faster near the surface in midstream and slower near the stream bed and banks due to friction. Average velocity across the whole stream is normally about 86% of the surface velocity you measure, so you can calculate average velocity by multiplying by 0.86.

Professionals typically use a current meter to measure velocity at different depths then calculate average velocity from these.

Current velocity is reported as distance per unit time (e.g., metres per second or m/s).

What does it tell us?

Velocity can be multiplied by stream cross-sectional area to calculate streamflow (see "Streamflow" section opposite). Also, if you have measured velocity on every monitoring visit, it can be used on its own, or with water level, as a relative measure of streamflow. Velocity indicates the potential for material (e.g., rubbish) to be transported downstream.

STREAMFLOW

Why is it important?

Streamflow (discharge) is the volume of water per unit time flowing past a point in the stream. It is a key measure of the size of the stream and of "state of flow" – how today's flow compares to the average.

The streamflow at the time of your monitoring visit is important for understanding the water quality on that day, as water quality depends strongly on streamflow. Therefore, it is important to estimate streamflow, or at least water level, on every monitoring visit.

The streamflow "regime" of a stream (median streamflow, mean streamflow and other statistics) describes how much a stream changes over time, usually in response to rainfall or snowmelt. A very changeable stream may be a more difficult habitat for aquatic plants and animals to live in than a more stable stream.

How is it measured and reported?

Water level measurements can provide a qualitative estimate of stream flow. Water level can be described as low, normal, slightly raised or high. If you are familiar with the stream, you can judge water level from your own experience. If you don't know the normal water level, then you can judge it in relation to perennial terrestrial plants and the stream banks. If the water is far below the perennial plants and the water is clearer than usual, the water level is low. If the stream is flooding over banks or over the roots of trees and shrubs, the water level is high.

Measuring streamflow (in m^3/s) gives more detailed information than estimating water level. For long-term monitoring sites, the best approach is usually to find a nearby site suitable for installing a staff gauge (similar to a vertical tape measure on a pole) to indicate water level on your stream visits. Water level can be converted to streamflow by developing and maintaining a rating curve.

Alternatively, you can estimate streamflow by measuring current velocity (see Current Velocity section) and cross-sectional area of the stream. Cross-sectional area is calculated by measuring water depths at 5-10 equally-spaced points across the stream channel and multiplying the average depth by the stream width.

Keep in mind that water level or streamflow measurements may collected (or estimated) by your regional council.

What does it tell us?

Streamflow on the day of (and 1-2 days before) your monitoring visit helps you to understand the water quality you measured that day, because most water quality indicators change with streamflow. For example, visual clarity is usually low at high streamflow and water is clear at low streamflow.

The streamflow in the days or weeks before your monitoring visit may help explain your results for periphyton, and may tell you whether it is a suitable time to sample macroinvertebrates.

STREAM HABITAT

Why is it important?

Stream habitat is defined as the whole stream environment including the stream bed, stream banks and land use in the immediate vicinity of the stream (riparian zone). It is formed by the interaction between several factors, including topography (shape of the land), geology, climate, and land-use.

The type and quality of this physical habitat have a significant influence on the stream plants and animals because each species needs a suitable "living space" to survive. Each species prefers different habitat conditions (e.g. some species prefer fast moving water, others quiet pools). Stream habitat provides:

- a place to live
- shelter from high flows
- protection from predators
- a place to lay eggs.

How is it measured and reported?

Stream habitat can be assessed several ways, ranging from visual observations to detailed measurements. In SHMAK we score eight different aspects of the stream bed, banks and riparian zone. Each aspect is assessed visually, by matching your site to descriptions that reflect a continuum of conditions from excellent to poor. This approach requires no equipment except a tape measure. Because stream habitat changes slowly under normal conditions, it only needs to be measured once per year. However, it is wise to re-assess after a large storm or new earthworks or construction activities.

What does it tell us?

Stream habitat assessments tell us about various human activities that may have degraded stream habitat (e.g., the removal of riparian vegetation, causing stream bank erosion, increased sedimentation, and smothering of fish habitat). Poor habitat conditions could cause a greater impact on stream life than poor water quality. Therefore, assessing habitat as well as water quality is necessary to interpret the results of biological monitoring.

Identifying which habitat features could be affecting stream health will help you set goals for restoring stream health. Monitoring stream habitat over time can also help you evaluate the success of your restoration efforts.

STREAMBED COMPOSITION

Why is it important?

The composition of the streambed (the type and size of particles that make up the bed) has a strong influence on stream life. Streambeds made up mostly of boulders and cobbles provide hard surfaces that stream bugs need to crawl on or under. So, they support a greater range and higher numbers of stream bugs than beds made up of fine sediments like silt. Cobbles and boulders also provide good habitat for native fish, creating spaces where the fish can shelter, feed and nest. Fine sediment (sand and mud) deposited on a stony stream bed can smother the hard surfaces macroinvertebrates like, fill up the spaces that fish use and clog their gills.

How is it measured and reported?

The most common method to estimate streambed composition is the Wolman walk (SHMAK Level 2). Randomly selected streambed particles are picked up, measured and the different size classes counted. A quicker method is a visual assessment (SHMAK Level 1), which can be made by walking up and down your stream reach and estimating the proportion of the streambed composed of each the categories given. This gives a rougher estimate than the Wolman method.

Streambed composition typically changes slowly so only needs to be monitored annually, ideally at the same time as you collect your benthic macroinvertebrate sample. But it can be useful to re-assess after a large storm or new development in the catchment.

Streambed composition is reported as the percentage of the stream bed that is covered by different categories and size classes of particles. Streambed particles range from mud or sand (<2 mm) to boulders (>25 cm). The stream bed may also be covered by leaves, wood, macrophytes (aquatic plants), concrete or bedrock.

What does it tell us?

Streambed composition tells us what sort of aquatic life we could expect to see (provided water quality and flows are sufficient). It also tells us both about the natural setting of a stream (catchment geology and topography) and about human influences. Streams in steep headwaters, in catchments with hard rock types, and those experiencing frequent large floods usually have beds dominated by large particles (cobbles and boulders). Streams in lowland, low-gradient valleys, in catchments with soft sedimentary rocks and/or having floods tend to have beds of fine sediment. A covering or build-up of fine sediment can also indicate soil erosion in the catchment or stream banks made worse by human activities such as earthworks or farming.

RUBBISH

Why is it important?

The pollution of fresh waters by household and commercial waste is a growing concern.

Rubbish:

- makes streams unattractive
- can make them unsafe for recreation
  - broken glass or discarded appliances can be sharp or dangerous
  - discarded chemical containers can leak toxic contaminants
  - some types (e.g. soiled nappies) can spread human pathogens
- can harm wildlife
  - animals can become trapped, strangled, or may eat some items.

Much of the rubbish is eventually transported downstream to estuaries or out to sea where it continues to harm the environment and pollute beaches.

How is it measured and reported?

A simple visual assessment can be made by estimating the amount of different rubbish types in and around a stream reach. In this SHMAK Level 1 method, several aspects of rubbish are scored on a scale of 1 to 8 and summed to give a final score out of 40:

Overall amount of rubbish	8	7	6	5	4	3	2	1
Threat to aquatic life	8	7	6	5	4	3	2	1
Threat to human health	8	7	6	5	4	3	2	1
Amount coming from on-site dumping	8	7	6	5	4	3	2	1
Amount accumulating from upstream	8	7	6	5	4	3	2	1
Total	**Score out of 40**							

For a more comprehensive understanding of what rubbish is there and where it is coming from, the rubbish tally method is used. In this SHMAK Level 2 method, rubbish items are identified and counted by material type (e.g., glass, metal, plastic) and use (e.g., food wrappers, metal appliances). They can then be weighed, and are removed from the site.

We recommend assessing rubbish once per year, or once each season if you want to determine the rate that rubbish accumulates in your stream reach in different seasons.

What does it tell us?

Assessing rubbish in streams can tell us where littering or dumping is occurring, can raise awareness of rubbish issues and show where better waste management strategies are needed. Be aware that the rubbish may have been dropped upstream or elsewhere in the catchment, and carried to your site by rain or wind. Rubbish surveys using the tally method can be linked with nearby coastal surveys of beach litter to help estimate the types and amount of rubbish exported out to sea. If you completely remove all the rubbish at your site and revisit the site regularly, then you can work out the accumulation rate (number of pieces per day) from your data, and compare accumulation rates at different stream sites.

CHAPTER 4
FIELD MANUAL

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INTRODUCTION

This chapter provides the information you need to use SHMAK in the field. It includes the instructions for monitoring each indicator, how often to measure and the equipment you will need. Before reading this chapter, you should have read through chapters 1-3 so you are comfortable with where, why, what and when you are monitoring. Chapter 3 in particular provides important information on each monitoring indicator outlined below.

Three categories of monitoring indicators are presented; water quality, stream life and stream habitat.

Water Quality – visual clarity, temperature, conductivity, nitrate, phosphate, *E. coli* bacteria.

Stream Life – periphyton (algae), macrophytes (aquatic plants), benthic macroinvertebrates (bugs), fish.

Stream Habitat – current velocity and streamflow, fine sediment deposition, habitat for aquatic animals, flow types, bank stability, riparian vegetation, shade, channel alteration, streambed composition, rubbish.

Identification Guides

SHMAK includes identification guides to help you in the field. These guides include photos and illustrations which will help you to identify various stream features along with stream plants and animals. The following NIWA field guides can be accessed on the NZ Water Citizens website:

- **Periphyton Identification Guide**
- **Benthic Macroinvertebrate Identification Guide.**

Training videos

Most of us are visual learners. So we've created some short videos that demonstrate the methods described in this chapter. The videos can be found on the website **www.nzwatercitizens.co.nz**.

HEALTH AND SAFETY

Safety is the most important element of any monitoring activity. Fill out a Health and Safety Plan before you first start monitoring. Read over your plan before every field trip and bring it to the field with you each time. Review the plan every year to check you have included any new hazards (e.g., a change in road or stream access). Here are some important things to think about when preparing your plan and as you monitor:

- **It's a team thing.** Monitor with at least one other person whenever possible. Teams of three or more people are better.
- **Phone in, phone out.** Let someone know where you are going and when you plan to return. Your contact person should have your contact phone number and know what to do if you don't come back at the agreed time.
- **First aid kit ready!** At least one member of the sampling team should have first aid training. Know any important medical conditions of team members.
- **How about the weather?** Dress for the conditions. Do not enter the stream if it is in flood. Monitoring some indicators during floods may be possible, but only from a bridge or using a pole sampler (see Chapter 2).
- **Be road safe.** Be sure your vehicle doesn't block traffic. Be careful crossing roads to access your sampling site.

Safety at the field site

High temperatures and physical activity can lead to heat stress. Remember Slip, Slop, Slap and Wrap (slip on a shirt, slop on sunscreen, slap on a sunhat, wrap on sunglasses) and drink plenty of water. Cold temperatures and rain can lead to hypothermia. Bring warm clothes, hat, rain gear and pack some dry clothes if it is cold or wet.

Safety in the stream

As part of your site selection process (Chapter 2) you should have identified one or more safe access points into the stream to use regularly. A low bank or a gravel bar on the inside of a bend are often good places. If you plan to take water samples from the stream bank during floods (using a pole sampler), identify a safe place to do this too.

- Be very careful when getting into the stream and walking in the stream.
- Some common hazards are unstable stream banks that may collapse, slippery stones in the stream, and deep pools or holes that are hard to see or filled with silt.
- Use a walking stick to steady yourself and to test for deep water or mud.
- Someone should always be on land ready to assist you if you fall, and a throw rope may be helpful.
- Do not attempt to cross streams that are swift and deeper than your knee.
- If the stream is in flood, do not enter the water.

Wear rubber boots and rubber gloves in streams suspected of having significant pollution (in reasonably clean streams, old sneakers, sports sandals or dive booties are fine). Chest waders are not recommended because they can become hazardous if you lose your footing. Wash your hands thoroughly with soap and water after monitoring or use antibacterial gel until you can reach a sink. This is especially important before handling food. Beware of potentially hazardous items, particularly in urban streams, such as broken glass or other sharp items, medical waste, building rubble, or discarded chemicals.


A.

B.


C.

Safety with chemicals

Several reagents used in the chemical test kits are considered hazardous substances. Please review the directions found in each kit carefully before using. Avoid contact between chemicals and skin, eyes, or mouth. Wear gloves (latex or nitrile) when doing these tests. If you do get a chemical on your skin or in eyes, flush with plenty of water. If you accidentally inhale or swallow some, call 0800 POISON. Store all chemicals away from children and pets, while avoiding extreme temperature fluctuations and direct sunlight. Properly dispose of all wastes from test kits. The chemicals described in this kit will break down naturally, so pour them onto soil or grass away from the stream and away from where people or domestic animals may make contact.

Safety with bacteria

Remember to safeguard your own health and the health of others when measuring bacteria in water, especially if you suspect your site may be contaminated with sewage or animal faecal pollution.

Polluted stream water can make you sick. Faecally-polluted water may contain enough disease-causing microorganisms to make you sick from direct contact. Wear gloves and wash hands thoroughly with soap before and after testing the water for bacteria.

***E. coli* plate + water sample = biohazard.** Once you have added your water sample to an *E. coli* plate, you should always treat it as hazardous. The bacteria will grow with the nutrients of the plate and in the warmth of the incubator, so even if the stream water had only a few nasty microbes at the start, the microbes will quickly multiply to dangerous levels. Keep the plate away from food preparation areas. Avoid direct contact with it by wearing gloves, sealing the plate with tape or keeping it in a resealable (e.g., Zip Lock) bag. You can "read" the plate without lifting its cover. We recommend that only adults handle the plates (and the water samples too, if you suspect they are polluted).

To dispose of your plates, add a tablespoon of bleach into a small resealable bag with no more than 10 plates. Seal it and put it in your household rubbish. Wear gloves and safety glasses for this step.

Keeping the environment safe – Biosecurity

As you will be coming into regular contact with streams and rivers, you have an important role to play in preventing the spread of invasive species such as didymo or aquatic weeds. Remember Check-Clean-Dry:

Check: for mud and plants on sampling equipment. Remove any that you find before you leave the site.

Clean: If you will be working on different streams in the same day, you will need to decontaminate your equipment after each sampling location. Rinse sampling gear with either a 2% solution of bleach (i.e. 20 mL of bleach for every 1 L of water) or a 5% solution of detergent or nappy cleaner. Either spray your gear with the solution or soak it in a bucket.

Dry: If visiting only one site on a day, air-dry your sampling gear completely. Complete drying might take 2-5 days.

If you are in areas where kauri grows, remember to clean your footwear to prevent spread of kauri dieback disease. Use the same rinsing solution as for decontaminating your gear.

TOP

Caption A. Dispose of wastes from test kits.

Caption B. Decontaminate your bacteria plates.

Caption C. Decontaminate your sampling gear.

GETTING READY TO MONITOR

The process for designing your monitoring programme and choosing sites is described in Chapter 2 of the manual. Here we assume you have chosen the monitoring site and just need to decide where at the site you will make the various measurements.

Re-check Health and Safety

Are there any known hazards here that you or someone else has identified previously? Are there any new hazards that have appeared, or are related to today's conditions? If so, note them on your Health and Safety Plan.

Choose your sampling area where you can safely step into the stream and wade to the measurement points.

Mark out your sampling reaches

A REACH is a length or section of stream that you define for a particular purpose (e.g., for monitoring particular indicators).

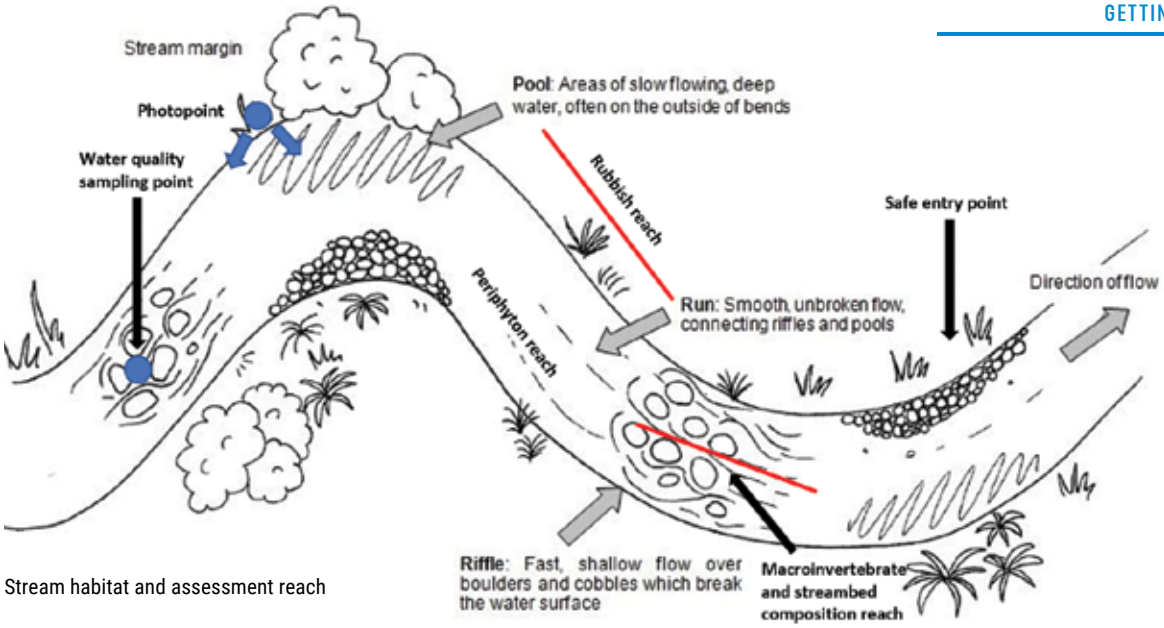
A RIFFLE is a shallow, fast-flowing area of the stream where the water surface is broken by ripples or small waves as the water flows over a rough bed.

A RUN is an area where the water is obviously flowing but the water surface is smooth or nearly smooth. Usually deeper than a riffle.

A POOL is a deep area of the stream where the water flows very slowly and the water surface is smooth.

The various assessments in SHMAK are done over different lengths (reaches) of stream. The shorter reaches should be within the longer ones (e.g. the periphyton reach of about 10 m within the stream habitat reach of about 50 m – see Stream Habitat diagram). Follow the steps below in order to mark out the longest reach first, then shorter reaches. If you are not doing the assessment for a particular step, go to the next step. Once you have chosen the longest reach you will use, anchor your tape measure at one end of the reach and run it along the stream bank to the other end. Keep the tape there until you have finished your day's monitoring.

It is best to monitor at exactly the same place each time you visit your site. So that you know where to monitor next time, draw a picture, take photos and/or write notes about your stream including key features like bends, boulders, logs, bankside trees, riffles, runs and pools, showing where your reaches are in relation to these features. Also write down the length of your longest reach.



Stream habitat and assessment reach

Stream Habitat diagram. An example of marking out reaches for stream habitat, rubbish assessment, periphyton, macroinvertebrates and streambed composition and locations for water quality sampling and photopoints in relation to flow types, landmarks and safe entry points.

1. Stream habitat assessment: We recommend a length of 50 m, or, if your stream is >3 m wide, a length of 20 times the stream width (depending on access, safety and how much time you have).
2. Rubbish assessment: Normally about 30 m long, where there is easy access along the stream bank.
3. Periphyton (for stony-bottom streams): Select a riffle, a run or both, according to whether one or both of these habitat types are typical of your stream.

   Or

   Macrophytes (for sandy- or muddy-bottom streams): Select a run that is representative of the stream.
4. Macroinvertebrates and streambed composition: For stony-bottom methods select a riffle, or a 10 m subsection of a riffle. For sandy- or muddy-bottom streams, choose a reach that includes all the common habitat types.
5. Water quality: Choose a place in the main current upstream of where you may have disturbed the water by wading.

Plan the order of measurements

The order that you take your measurements can be important. Some measurements involve wading in the stream, which disturbs the bed sediment, while others need an undisturbed stream to get good results. On the monitoring trips when you are measuring all indicators, measure in this order:

1. Water quality
2. Stream life (periphyton, aquatic plants and macroinvertebrates)
3. Stream habitat.

If you have several people taking measurements at the same time, spread out and ensure that those measuring water quality are upstream of the others.

RECORD KEEPING

It is important to keep records of your project and your monitoring sites, including detailed information of each visit to your site.

Project and site information

Before you begin monitoring it is important to record some basic information for your monitoring project and sites within the project. Project information includes the project title, description, location, purpose or goal, and names of members. On the NZ Water Citizens website you can choose whether to share your project publicly or keep it viewable by yourself or your group only. You can also add web links and attach other information.

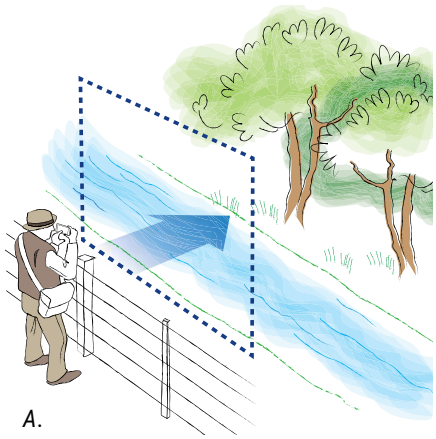
Site information includes:

1. Site name or code – a unique name that describes where on the stream the site is, for example Hutt River at Melling Bridge.
2. Stream/river name – for example the Hutt River.
3. Site coordinates – these are the map coordinates for the site, taken from a topographic map (such as the map on www.nzwatercitizens.co.nz), a GPS or your phone GPS.
4. REC reach – REC is the River Environment Classification, a web-based map that gives an identification number for each stream reach in New Zealand. You can find the REC reach number by locating your site on the map in www.nzwatercitizens.co.nz.
5. Type of site – whether this is an impact site, control site, reference site, input or output site (see Chapter 2 or glossary for definitions of these).

Site visit information

Every time you monitor, fill out a SHMAK field form to capture important information on site and weather conditions that might affect the water quality or stream life (such as livestock or birds in the channel), and water surface features (foams, scums) and odour. The field form is available on the NZ Water Citizens website and includes the following headings:

1. Date and Time – the date and time when the assessment was made.
2. Monitoring team – the names of the people doing the assessment on this day.
3. Photographs – Take a photograph of the site from a photopoint and photograph anything else that may be useful for interpreting your water quality results. If you are going to multiple sites, it can be helpful to first take a photo of your datasheet (with site name and date filled in) so you can associate the photos with that site.



A.

B.

C.

TOP

Caption A. Set location of photopoint

Caption B. Use a distinctive object.

Caption C. Note features at edge of photo.

4. Weather conditions – a description of the weather conditions on the day of collection (weather now) and the rainfall in the past 48 hours.
5. Water level – Record as low, normal, slightly raised or high. This is an estimate of streamflow. To measure streamflow in m³/s, see *Current velocity and Streamflow* section later in this chapter.
6. Length of reach – river width, maximum depth.
7. Main land use in catchment, and upstream catchment disturbances – main land use might be (for example) native forest, dairy farm or urban. Catchment disturbances could include earthworks, a stormwater discharge, forest harvest, etc.

Photopoints

Photopoints are a simple way of recording important information about your site and how it changes. They are a series of photos taken on many occasions over a period of time:

- at the same location
- in the same direction (or "bearing"), and
- with the same "frame" (how wide the view of the camera is).

How to make a photopoint:

Set the location: set up a distinctive object (e.g. a Y-post or waratah), or find an existing one (e.g. a fencepost) on which to place the camera each time. Or locate yourself in relation to a distinctive object that is unlikely to move, like a tree. Mark the object you place your camera on, write notes about how to find it and record the GPS coordinates.

Set the direction: record the bearing (compass direction) and note a distinctive feature that is included in the photo.

Set the frame: note features that are near the edges of the photo.

Site health check

If it's your first time at the site, you will need to find out a few things about it. If you have been there often, it's useful to see if it has changed since your last visit. Think SOSMART to run a quick "health check" and help pick up any particular issues in your stream:

Smells. Make sure the smells you record are coming from the stream itself and not somewhere nearby. It might help to take a close sniff from a bucket of water pulled from the stream.

Obstructions. Anything that is restricting water flow.

Stream bed. Record anything that is on, covering, or smothering the streambed.

Margin or bank. A healthy stream has thick vegetation (trees or shrubs) growing on the bank. Problems with the stream bank or margin include trampling by livestock, other erosion or bank collapse, rubbish, paving or other artificial materials, or lack of tall vegetation.

Appearance of the water. A healthy stream should have fairly clear water except for a short time during high flows. Discolouration or murkiness in the water at normal or low flows may indicate problems.

Rate of flow. Although some streams are naturally slow flowing, slow flow may create a difficult habitat for stream animals to live in (e.g. because of low oxygen), and can be caused by various human impacts such as building weirs or dumping rubbish.

Top surface of water. Some pollutants (e.g. detergents) float or create foams that float on the water surface, though foams could also be from natural organic substances. "Blooms" of algae often create floating green swirls.


A.


B.


C.

HOW TO COLLECT A WATER SAMPLE

Some of the tests require you to collect a water sample for testing on the stream bank (e.g. nitrate or phosphate) or at home *(E. coli)*.

Before you go

Ensure you have a clean bottle or container. Use the 100 mL container in your SHMAK kit or a mineral water bottle from a supermarket. Clean it by washing with phosphate-free detergent, rinsing at least five times in tap water and air drying. If collecting for *E. coli* your container should also be sterile (see *E. coli* method for how to sterilise). Bring a back-up container in case you accidentally contaminate the inside of your first one.

TOP

Caption A. Collect sample in main flow.

Caption B. Put lid on underwater.

Caption C. Put in a chilly bin.

Sampling for nutrient tests

1. Collect samples in the main flow of the stream (for small streams, this is usually mid-channel), just below the water surface.
2. Approach your sampling location from downstream, disturbing the bottom sediment as little as possible. Always face upstream to collect your samples or take measurements, to avoid any stirred-up mud from the streambed getting in your sample.
3. Remove the lid from the container just before sampling. Avoid touching the inside of the container or the lid. If you accidentally touch the inside of either, use your back-up container or triple-rinse with stream water.
4. Rinse your container by tipping it upside down and pushing it about 20 cm below the water surface. Turn it upright so it fills. Tip that water out and repeat.
5. Repeat again but keep the water sample. Put the lid on while the container is still underwater.

Sampling for *E. coli*

Follow the same steps as for nutrients. The differences are:

1. Water samples collected for faecal bacteria testing are easily contaminated. So:
   - use a separate sterile container
   - ensure your hands are clean
   - avoid touching any part of the inside of the container or cap.
2. Bacteria die off rapidly if exposed to sunlight even for a few minutes. So:
   - immediately wrap the sample container in aluminium foil to keep it dark
   - place it in a chilly bin with a frozen ice pack to keep it cool.
3. *E. coli* samples should be processed within 24 hours of sampling (ideally on the same day).

Sending a water sample to a professional lab

1. Plan ahead. Ensure you have the correct water sampling container for the test (ask the laboratory staff). Make sure the timing will work for you and the lab.
2. Label the container (site name, date, time, your initials).
3. Collect the sample as described above. Transport it in a chilly bin (cooler) with an ice pack.

VISUAL CLARITY

How to use a black disc viewer

Number of people: 2

One to hold the disc, the other observing.

Equipment:

Black disc on pole

Underwater viewer box

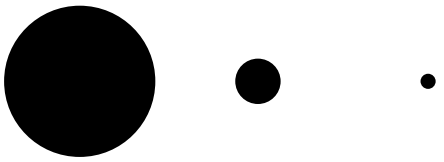
Measuring tape

Instructions

1. Your partner holds the black disc just below the water surface, upstream or across the stream from you (the person with the viewer box).
2. Attach the tape measure to the black disc. Pull the tape tightly and wait for any disturbed sediment to settle.
3. Look into the viewer box, with the top of the viewer box snug against your face. Allow time for your eyes to adjust. You should see the black disc.
4. Walk carefully backwards downstream, or across the stream. Keep the viewer box snug against your face and continue to hold the measuring tape. When the disc just disappears from your sight, record the distance (y1) between the viewer box and the disc.
5. Walk slowly towards the disc until it just re-appears and record this distance (y2). Visual clarity is the average of these two distances:

$$\text{Visual clarity} = \frac{y1+y2}{2}$$

Which black disc size to use?



Size of disc	200mm	60mm	20mm
Use if clarity is	>1.5 m	0.5 to 1.5 m	<0.5 m

How to use a clarity tube

Number of people: 1 or 2

Easier with 2 people holding the tube.

Equipment:

Clarity tube with pair of magnets

Bucket (2 L or larger)*

* not supplied with SHMAK

Instructions

1. Collect at least 2 litres of stream water (clarity tube is about 1.4 litres) in a bucket. The water should come from the main stream flow and contain no streambed sediment stirred up as you walk.
2. Remove the black cap from the tube and pour the water sample to the very top of tube (to keep air bubbles as small as possible).
3. Add the aquarium magnet to the inside of the tube and the matching aquarium magnet to the outside of the tube. Recap the tube.
4. Place the tube horizontally on a stable surface (e.g. a fence post) or have a second person hold the end of the tube. If there is a small bubble in the tube, make sure it is at the cap end, not the viewer end.
5. Starting with the aquarium magnet near you, look at it through the viewing window. Then slowly move the magnet away from you until it is no longer visible. Note the distance to the near end of the magnet (y1) using the marks along the side of the tube.
6. Now slowly move the magnet towards you until it just re-appears. Note this distance (y2). Visual clarity is the average of these two distances:

$$\text{Visual clarity} = \frac{y1+y2}{2}$$

Note: If the clarity is less than 0.1 m you will get a more accurate measurement by diluting the stream water with clean (e.g. tap) water and dividing the measured clarity of the diluted sample by the dilution factor. For this you will either need to bring tap water or you will need to take your water sample home.

B.

C.

TEMPERATURE

How to use a thermometer

Number of people: 1

Equipment:

Thermometer (or conductivity meter with built-in temperature sensor)

Instructions

1. Measure temperature in the main flow of the stream. Leave the thermometer or conductivity meter in the water until the reading stabilises (at least 1 minute).
2. If possible, try to read the temperature with the thermometer bulb beneath the water surface. If not possible, you can fill a bucket with water and measure the temperature of the water in the bucket.

How to use a temperature logger

Number of people: 1

Equipment:

Temperature logger

Waratah (Y post) or similar for attaching logger*

Sledge hammer*

Wire or thick cable tie*

Wire cutters or pliers*

Mobile phone with app installed*

* not supplied with SHMAK

Instructions

1. Follow the manufacturer's instructions for setting up and launching the logger and downloading the data. Set the logger to record every 30 minutes.
2. Hammer a waratah (Y post) into the stream bed, near the stream bank where there is definite water movement but not in the main current (where it could get pulled out or damaged during high flows). Leave one of the waratah holes near or just above the water surface.
3. Loop the cable tie or wire through one of the holes in the waratah and attach the temperature logger using wire or a thick cable tie. Check the wire/cable tie on each visit and replace if you see signs of wear or rust.
4. Adjust the height of the waratah so that the waratah is at least 50 cm into the streambed and the logger will be below water level during the lowest flows. It should be well above the streambed so it doesn't get buried in a flood. If possible, keep it shaded from direct sunlight.
5. Mark the place you have installed it (or photograph and write notes on its location) so you can find it easily next time.
6. If there is something stable in the stream that will not get washed away in a flood, e.g. submerged tree roots or a large log, you can attach the logger to this instead of a waratah. Just make sure the logger is near the stream bank, in the stream flow and will be below water level during the lowest flows.
7. Download the data to your mobile phone every time you visit your site, following the manufacturer's instructions.

TOP

Caption A. Measure temperature in stream.

Caption B. Hammer waratah into stream bed.

Caption C. Attach logger.

A.

CONDUCTIVITY

How to use a conductivity meter

Number of people: 1

Equipment:

Conductivity meter

Sample container

Conductivity standard solution

Spare batteries*

* not supplied with SHMAK

Instructions

1. Rinse a water sample container or bucket with stream water and fill with stream water.
2. Remove the cap from the bottom of the probe. Switch on the instrument and check it reads zero in air.
3. Place the probe into the stream water sample. Ensure that the water does not go above the grey line, as this will cause leakage into the instrument and corrosion of the circuit board.
4. Allow the reading to stabilise (this should take only a few seconds), then record the conductivity. Switch off when finished! The batteries are expensive to replace.

How to recalibrate your meter

Conductivity meters get gradually higher or lower over time so check the accuracy of your readings every visit.

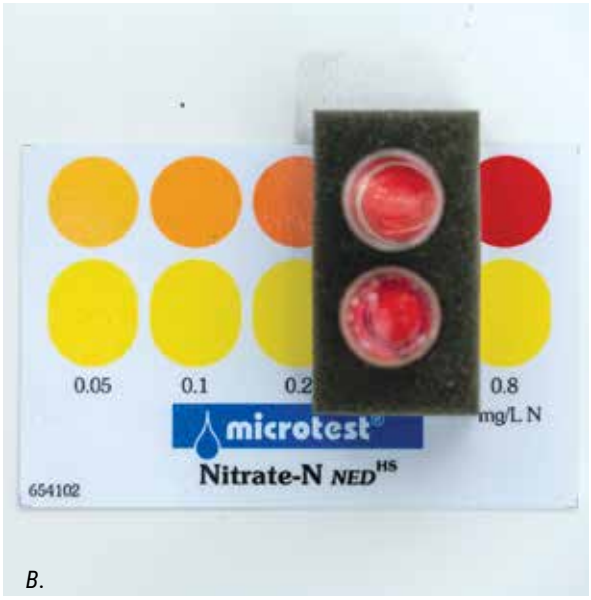
1. Clean the tip of your meter by washing in tap water and wiping with a tissue.
2. Dip the meter into the conductivity standard solution in your kit and note the reading. If it doesn't match the label on the bottle you will need to recalibrate your meter.
3. Adjust the meter so it shows the correct reading. The instructions for adjusting your meter should be in the box or pouch that the meter came in.

The conductivity standard solution in your kit should be good for 3-4 years, provided you keep it in a cool dark place with the lid tightly on, and only ever put the cleaned tip of your conductivity meter into it. After this time, you may need to replace it. Ask your regional council or local high school for help with this.

TOP

Caption A. Measurements can be taken in a bucket of water.

A.



B.

NITRATE

Health and Safety: The chemical powder used in this test (Reagent B) is a hazardous substance. Wear gloves and wash your hands after using the kit.

How to use the Aquaspex Microtest® Nitrate-N kit

Number of people: 1

Equipment:

Aquaspex kit, including:

2 vials (5 mL)

Reagent A (liquid) and B (powder)

Small scoop

Foam block

Colour comparator card

Other items:

Disposable gloves

Water sample container

10 mL syringe

Instructions

1. Collect a water sample following all the steps in 'How to collect a water sample' (*page 52*).
2. Rinse both vials several times with the sample water. Fill each vial to the 5 mL mark (using a syringe to measure 5 mL is easiest).
3. Place one vial in the foam block. This is your reference vial. Do not put the lid back on.
4. To the second vial add 6 drops of Reagent A, cap the vial and turn it over several times to mix. This is your measurement vial.
5. To the same vial add 1 level measuring spoonful of Reagent B (using the scoop provided). Cap the vial and shake vigorously for 60 seconds.
6. Allow to stand for 3 minutes for full colour development. Mix occasionally by turning the vial over several times. The sample will gradually develop a pinkish colour if nitrate is present.
7. After 3 minutes, remove the cap and place the vial in the foam block.
8. Place the foam block on the colour comparator, with the reference vial in the top position and the measurement vial in the bottom position (both with their caps off). Looking from above, move the foam block across the colour fields until you get the best colour match between the two solutions. Read the concentration of nitrate-nitrogen (in mg/L) at the bottom of the card.
9. If your reading seems darker than 0.8 mg/L, dilute it 1:1 (or a lower ratio) with distilled water (available from automotive stores, e.g. Repco). You can use tap water or bottled water for diluting provided you ensure it measures 0 mg/L nitrate-N using the Aquaspex kit.
10. To dispose of your sample, pour it onto grass or soil at least 5 m away from the stream. Rinse the vial with clean water before putting it away.

TOP

Caption A. Keep the reference vial in top position.

Caption B. Move the foam block until colours match.

A.

B.

PHOSPHATE

Health and Safety: The chemical powder used in this test (Potassium disulfate) is a hazardous substance. Wear gloves and wash your hands after using the kit.

How to use a Hanna Instruments Checker

Number of people: 1

Equipment:

Hanna Instruments Phosphate Checker LR, including:

Colorimeter

Batteries

2 vials

Chemical powder (reagent)

Other items:

10 mL syringe

Phillips head screw driver*

Scissors

Lint free cloth or tissue*

Disposable gloves

Stopwatch or cell phone timer*

Water sample container

Blue tack*

Funnel

Picnic table* (optional)

Spare batteries*

* not supplied with SHMAK

Before you start:

Before using the colorimeter for the first time you will need to insert the batteries. This requires a small screwdriver to open the battery compartment. Check the expiry date on the package to ensure the reagents haven't expired. You can order replacement reagents from the company using the contact details in the Appendix.

Instructions

1. Collect a water sample following the steps in '*How to collect a water sample*' (*page 52*).
2. Use the instructions below instead of the instruction card in the box.
3. Put on a pair of gloves to protect your hands from contact with the chemical powder.
4. Pour the chemical powder (reagent) from the foil packet into vial 1 and add 10 mL of the water sample. This is your measurement vial. See the tips for adding the chemical powder below. Screw the cap on the vial.

Tips for adding the chemical powder (reagent)

a. Place a funnel into the mouth of vial 1 (the sample vial). You may want to wrap a thin bead of blue tack around the neck of the funnel to keep it in place.

b. Stand the reagent packet upright on a hard surface and tap it gently so all the chemical powder falls to the bottom of the packet.

c. Cut around two sides (following the dotted line on the packet) with a pair of scissors.

d. Pinch the open corner of the packet so it forms a crease in the shape of a V.

e. Place the V over the funnel and gently tap or 'flick' the other end of the packet with your finger to move the chemical powder along the crease and into the vial (Fig.1).

f. Fill the syringe with 10 mL of sample water and gently squirt this water into the funnel so any powder stuck on the sides of the funnel is washed into the vial (Fig.1). When finished, the vial should be filled to the 10 mL mark.

g. Don't worry if you have spilled a small amount of the chemical powder. A small loss should not affect the results.

TOP

Caption A. Add reagent to vial taking care not to spill.

Caption B. Place vial in checker.

DRAFT

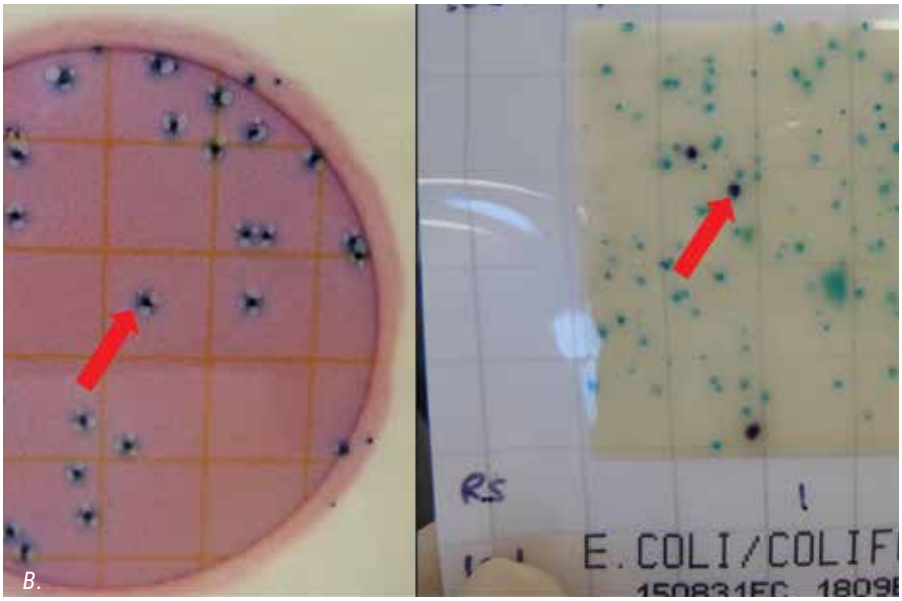

A.

TOP

Caption A. Use syringe to remove powder stuck to funnel.

5. Start the stopwatch and gently shake vial 1 for 2 minutes to dissolve the powder.
6. When the 2 minutes is over, stand vial 1 on a flat surface.
7. While the stopwatch is reading between 3 and 4 minutes, turn on the colorimeter by pressing the black button on the front. When the display shows “Add”, “C.1” with “Press” blinking, the meter is ready.
8. Fill vial 2 (the reference vial) to the 10 mL mark with sample water. Wipe any water drops off the outside, insert the vial into the colorimeter and close the lid. Press the black button. When the display shows “Add”, “C.2” with “Press” blinking, the meter is zeroed.
9. Wipe any water drops off the measurement vial. If there are bubbles inside, tap the vial gently.
10. When the stopwatch reads 5 minutes, insert the sample vial into the colorimeter, close the lid and press the black button to measure the phosphate in your sample.
11. Write down this number. It is the concentration of phosphate in mg/L. If you want the concentration as phosphate-P, then multiply this value by 0.326.
12. Switch off the colorimeter by pressing the black button again. It will turn off automatically in 2 minutes.
13. To dispose of your sample, pour it onto grass or soil, at least 5 m away from the stream. Rinse the vial in clean water before putting it away, as the chemical will etch the glass.

A.



B.

E. coli BACTERIA

Health and Safety: Bacteria multiply in a warm environment like the incubator in this test, so treat each plate as hazardous after adding your sample to it. For safety, seal each plate with tape or put in a resealable bag. Wear gloves and wash hands thoroughly with soap or hand sanitiser before and after water analysis.

Storage: Refrigerate unopened *E. coli* plates. Once opened, keep *E. coli* plates in a tightly sealed package to prevent condensation from building up on the plate. Use within a year.

TOP

Caption A. Put plates in waterproof box in incubator.

Caption B. Colonies are blue with gas bubbles (Petrifilm™) or indigo/purple (MCM).

How to sterilise your equipment

Three items need sterilising so you don't contaminate your sample with bacteria from elsewhere. **These are the water sample container, pipette, and tweezers.**

Sterilise your equipment before your first use, and again after each use. Do this by washing in a tub of water with dish detergent and wiping with a soft cloth. After the detergent wash, rinse each item at least five times in tap water, then twice with water that has been boiled and cooled in a jug. Air-dry each item indoors on a clean surface (ideally in direct sunlight) and store dry in a ziplock bag.

The most important ways to lower the risk of contamination are to rinse the sample container several times in stream water before you take your sample, and "pump" the pipette several times in the water sample before removing 1 mL for analysis.

Note: The following instructions are for MCM *E. coli* plates. If using 3M™ Petrifilm™ *E. coli*/ Coliform plates, also consult the additional instructions in the appendix.

How to use MCM *E.coli* plates

Number of people: 1

Time required:

<5 minutes to sample, 10-15 minutes to add water sample to *E. coli* plate, 24 hours to incubate, 5 minutes to read the plate.

Equipment:

Water sample container (sterile)

Aluminium foil*

Chilly bin (for incubator and transporting sample)

Ice pack*

Disposable gloves

Pipettes (sterile)

E. coli plates

Syringe with connector tubing

Filter cup with filter paper

Plastic sandwich box

Tweezers (sterile)

Permanent marker pen*

Resealable bag

Jar lid (wider than *E. coli* plate)*

Aquarium heater

Bleach*

* not supplied with SHMAK

3.

5.

Instructions

Collecting a water sample

1. Collect a water sample following the steps on page 52. Immediately place the sample out of direct sunlight by wrapping in foil and placing in a dark box (or chilly bin). Bring the sample container home within 1 hour or keep the sample cool, e.g. in a chilly bin with a frozen ice pack.

Processing your sample at home

2. If you can't process your sample immediately, put in the fridge for up to 24 hrs.
3. Warm up your incubator. Check the temperature is stable at 33-37 °C before putting your sample inside.
4. Allow your sample plates to come to room temperature (10- 15 minutes). Check the expiry date on the package.
5. Label the top flap of the plates with the site name, date, sample volume used and the initials of the tester. Place the plate on a flat surface.

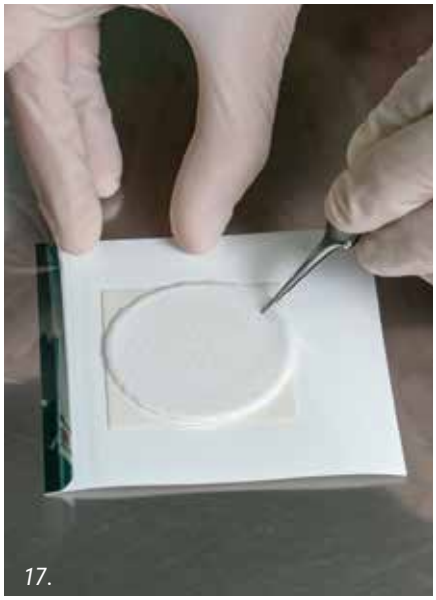
How many plates should I prepare?

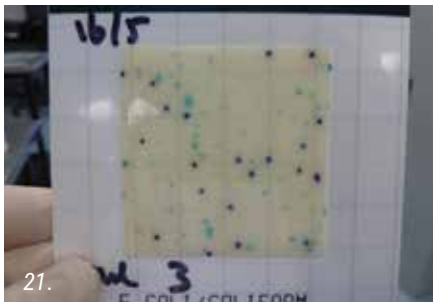
You will get the best results if you end up with 20-80 colonies on the plate. If you expect your site has high levels of *E. coli* (>500 cfu per 100 mL) then the **Direct Plating** method will give the best results. If you think your site has fairly low numbers of *E. coli* (<500 cfu per 100mL) and the water level was not high when you sampled, then the **Filtering** method will give better reuslts. Until you get good at predicting *E. coli* concentrations, we recommend that you prepare two plates per water sample (one for each method).

Direct Plate Method

6. Turn the sample container over several times to mix.
7. Insert the pipette in the water sample, "pump" it several times, then measure out 1 mL.
8. Lift the clear film and add the 1 mL water to the plate.
9. Carefully roll the top flap back down, taking care to avoid trapping air bubbles.
10. If the water doesn't spread all around the gel by itself, you can tilt it slightly to help it spread.


15.


17.



21.

Filtering Method

11. Pre-wet the MCM plate fabric using 1 mL of sterile water (tap water boiled and cooled in a jug), measured using a sterile pipette
12. Moisten the filter paper by adding a few mL of sterile water to the filter cup using a sterile pipette.
13. Turn the sample container over several times to mix the sample thoroughly.
14. Decide what volume of sample you will use (Table 1). Pour this volume of sample into the filter cup, using the marks on the side of cup. If you want less than 20 mL, use a sterile pipette which can measure up to 3 mL at a time. Keeping the filter cup level, attach the syringe to the underside of the cup.
15. Draw the sample through by pulling down the syringe plunger and discard the filtered water. If you need to filter more than 50 mL of sample, you will need to repeat this step.
16. Rinse the sides of the filter cup by adding sterile water with a sterile pipette.
17. Remove the cup from the filter apparatus and lift the filter paper off the base using sterile tweezers. Place the filter paper on the MCM plate face up.
18. Store unused sample water in the fridge for up to 48 hours. If test results are unclear, you can repeat it on the unused sample.

Table 1. Volume of water to filter to get 20-80 colonies on your *E. coli* plate. This should be taken as a guide, e.g. streams in urban areas, near leaking septic tanks or with stock access may have more *E. coli* than expected from this table.

Visual clarity	Volume of water to filter
> 1.5 m	50 – 100 mL
1 m to 1.5 m	20 mL
50 cm – 1 m	10 mL
< 50 cm	Direct plating method

Incubation and Processing

19. Tape (or staple) the top flap of the MCM plate down and put in the incubator. If using a water bath incubator, place the plate in a resealable bag and place a jar lid (wider than the plate) over it to keep the bag from pressing on the gel. Seal the bag. Place the bag in a plastic box and float it on the water surface.
20. Note the time and leave the plate in the incubator for 24 hours.
21. Remove the plate from the incubator and the bags and count the number of purple-blue dots (*E. coli* colonies). You do not need to lift the flap (and should not, as **the plate is now a health risk).**
22. Divide the number of colonies by your sample volume (in mL), multiply by 100 and record this number (as *E. coli* cfu per 100 mL) in your data sheet. For example:

Count	Sample volume (mL)	Result (cfu per 100 mL)
50	1	5000
20	10	200
10	40	25
7	100	1

Disposal

23. Place a teaspoon of bleach onto the surface of the plate and allow to sit for at least 5 minutes. Place in a water tight bag and discard in household rubbish.

PERIPHYTON

Health and Safety. If the water is turbid (cloudy) and you can't see the streambed or if flows are high and you can't safely enter the water, then do not attempt a periphyton assessment. Just note on your sampling form why no periphyton assessment was done.

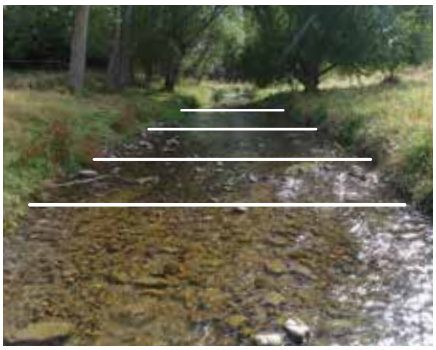
Where to do your periphyton assessment?

The periphyton assessment is designed for "wadeable" streams with a gravel or cobble bed. These are the environments where periphyton impact most on human values and aquatic ecology. Focus on riffle or run areas and avoid the pools. Record whether your transect was in a riffle or a run, as this information will help you to interpret your data later on. If you want to compare two or more sites, try to compare riffles with riffles or runs with runs. Carry out assessments in stream reaches with less than 70% shading, unless you specifically want to assess periphyton in a shaded stream or are comparing with another shaded site. As a rule-of-thumb, shading is less than 70% where riparian trees are shorter than the width of the stream channel (including any gravel banks).

Locating your observation points

You will need a total of 10 survey points at minimum, or 20 for higher accuracy. Keep the same general location of your observation points on each monitoring visit. Space the observation points evenly along the path or transect so that they include a range of water depths and velocities.

Very small (<2 m wide), wadeable stream: estimate periphyton cover at points along a zig-zag path that extends to the water's edge on each side.



Shallow (2–15 m wide) wadeable stream: 2–4 transects from bank to bank. Estimate periphyton at 3–5 evenly-spaced points across each transect, including the middle and (near) the sides, e.g., at 10%, 30%, 50%, 70% and 90% of stream width.

Larger (>15 m wide) and/or partially unwadeable streams (deeper than about 0.6 m): 2–4 transects extending partway across river. Space observation points evenly across transect.

Estimating periphyton coverage

Estimating the percentage cover of periphyton at a site takes some practice. First you need to know how to identify the types of periphyton assessed in this method (filamentous and mat-forming algae) and distinguish them from moss.

There are two main methods – the stone method (Level 1) and the viewer method (Level 2). They may give slightly different results, so it is best to stick with one method. The stone method is a little quicker than the viewer method, does not require any equipment, and may be better if visibility is poor underwater. In the stone method, you estimate the percentage of the top surface of each stone (the surface exposed to light) covered by each of the periphyton categories and moss.

The viewer method is more accurate than the stone method and more representative of the whole study area. In this method, you estimate the percentage of the streambed in your field of view that is covered by each of the periphyton categories and moss. For a viewer you can use your black disc viewer (with the mirror removed so you can see through the bottom window). A bathyscope is better than the black disc viewer because it has a wider field-of-view, but it is quite bulky to carry. It is not included as part of SHMAK, but can be purchased from marine stores (see Appendix).

Identifying periphyton

Periphyton is classified in broad categories based on growth form: thin films, sludge, mats, and filaments (see examples below). Moss, although technically not periphyton, is included in the periphyton assessment as it sometimes grows on rocks on the streambed. Macrophytes (aquatic plants) are assessed separately.

More details on identifying periphyton are given in the *Periphyton Identification Guide*.

Categories of periphyton (left to right from top): thin films, sludge, mats, filaments (out of water) filaments (underwater), moss.

How to use the stone method (Level 1)

Number of people: 1

Easier with second person recording.

Equipment:

Bucket (if collecting benthic macroinvertebrates at the same time)

Periphyton Identification Guide

Instructions

1. Working from the downstream end of your site, move out to the first point on your path or transect. Without looking at what is there, bend down to touch the sediments of the streambed and pick up the first stone that you touch provided it is >4 cm wide. If it is <4 cm wide, pick up the nearest 4 cm-wide stone instead.
2. If you are collecting benthic macroinvertebrates (bugs) using the Stone Method place the stone in a bucket to prevent losing any bugs, and bring it to shore.
3. Examine each stone carefully and identify if there is filamentous algae, mat-forming algae or moss present using the Periphyton Identification Guide. Note: mat-forming algae does not include Microcoleus or didymo. These are recorded separately.
4. Now determine the percentage of the rock's upper surface (the surface exposed to light) that is covered by each type of periphyton and moss. Estimate to the nearest 5% (or to the nearest 1% if cover is less than 5%). Record this on your data sheet.
5. Repeat this at the 10 (or 20) observation points along your transects. Try to evenly space your stones along the transect so you are not selecting stones with more (or less) algae.
6. Record if your assessment was done in a riffle or a run, whether you found detached mats of *Microcoleus* on or near the shore, and the main colour of the periphyton mats and filaments.

How to use the viewer method (Level 2)

Number of people: 2

1 to observe, 1 to record.

Equipment:

Black disc viewer (with mirror removed) or bathyscope

Instructions

1. Starting at the downstream-most transect, move out to the first point on the first transect to be sampled.
2. Facing upstream, hold the viewer with the bottom glass below the water surface. Aim to hold the glass about 20 cm above the streambed, although this distance will vary with depth. If the water is clear and the light is good, this should give you a clear view of the stream bed.
3. Identify if there is filamentous algae, mat-forming algae or moss present using the Periphyton Identification Guide. Note: mat-forming algae does not include *Microcoleus* or didymo. These are recorded separately. Estimate percentage cover of each periphyton type in your field of view to the nearest 5% (or the nearest 1% if cover is less than 5%).
4. Repeat this at the 10 (or 20) observation points along your transects. Try to evenly space your observations along the transect so you are not biased towards points with more (or less) algae. You can move the black disc viewer around at each observation point to get a wider field of view if this helps.
5. Record if your assessment was done in a riffle or a run, whether you found detached mats of *Microcoleus* on or near the shore, and the main colour of the periphyton mats and filaments.

How to assess *Microcoleus* for human and animal health risk

Number of people: 1

Equipment:

GPS* (if needed)

Tape measure

* not supplied with SHMAK

If you found *Microcoleus* in your periphyton assessment, you can use the following method to assess whether it poses a threat to human and animal health.

Instructions

1. Mark out a reach for your assessment. If access along the bank or shoreline is easy, make your reach length at least 20 times the width of the stream, up to a maximum of 100 m long. Record the length of the reach and GPS coordinates or landmarks that mark the upstream and downstream ends.
2. Walk the length of the reach along the bank (or both banks if you can), and record whether or not you find detached mats of *Microcoleus* floating by the shoreline or washed up on shore.
3. Either on your bankside walk or standing on a bridge where you can see the entire reach, estimate the % cover of the entire streambed surface that is covered by *Microcoleus*.

A.

B.

MACROPHYTES

How to do a visual assessment

Number of people: 1

Easier with second person recording

Equipment:

30 or 50 m tape measure

1 m ruler or measuring stick (optional)*

* use SHMAK one or make your own

Setting up the monitoring cross sections

Run the tape measure along 20 to 50 m of the stream bank in a "run" area (where the water is definitely moving but a little deeper and smoother than a riffle). You will assess macrophytes at up to 5 evenly spaced cross sections within this area. Decide on the spacing between your cross sections (e.g. every 5 m starting at the 5 m mark on the tape measure).

Instructions

1a. If the water is clear enough to see the bottom and the stream is less than about 3 m wide, then you can assess macrophytes while standing on the bank.

1a.1 At each cross section, picture a 0.5 m wide band across the stream. Estimate the percentage of the water surface that is covered by macrophytes (to the nearest 10%). Macrophytes that don't reach the water surface are recorded as 0% surface cover.

1a.2 Picture a volume of water defined by the 0.5 m band across the stream and extending from the water surface to the streambed. Estimate the percentage of that volume that is occupied by macrophytes (to the nearest 10%). See Fig. page 70 for help.

1b. If you can't see the bottom clearly all the way across, or the stream is wider than 3 m, you will need to get into the stream.

1b.1 Choose 3-5 points (depending on the stream width) evenly spaced across the stream. At each point picture a square on the water surface 0.5 m x 0.5 m wide. Estimate the percentage of the water surface that is covered by macrophytes (to the nearest 10%). If the macrophytes don't reach the water surface, record 0% surface cover.

1b.2 Then picture a column of water under this square going down to the streambed. Estimate the percentage of that column that is occupied by macrophytes (to the nearest 10%). See Fig.2 page 70. If you can't see to the bottom, you may have to feel around to make your estimate.

2. Assess macrophyte water surface cover and clogginess

2.1 The "macrophyte water surface cover index" is the average of all the bands or columns for percentage of water surface. The "macrophyte clogginess index" is the average of all the bands or columns for percentage of water column.

TOP

Caption A. You may need to get into the stream to assess macrophytes.

Caption B. Create a transect along a run section.

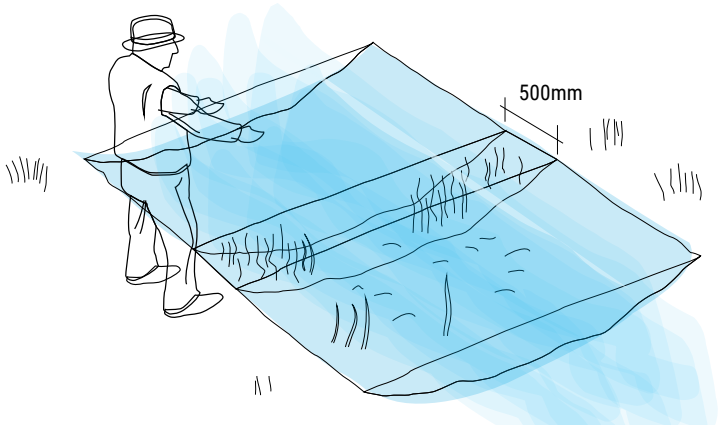


Fig. 1 If the stream is narrow and the water is clear, picture a 0.5 m wide band across the stream. Estimate % of water surface and % of water volume occupied by macrophytes within this.

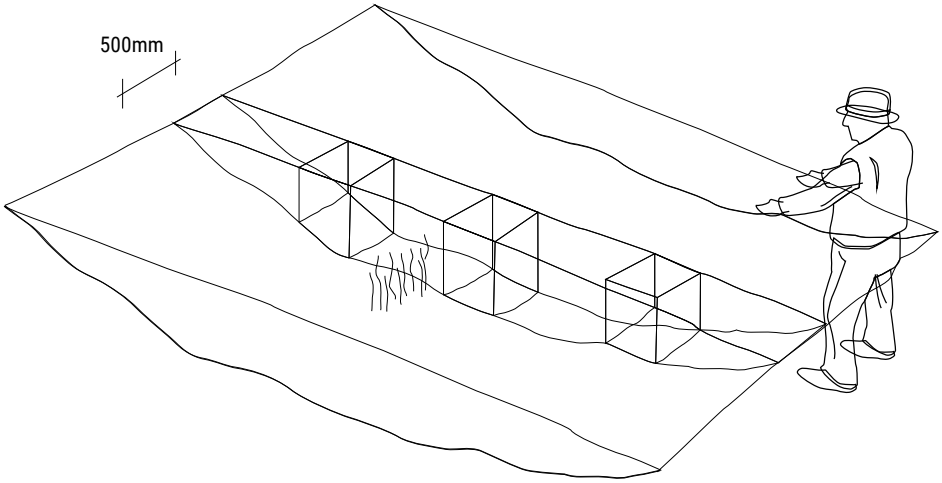


Fig. 2 If the stream is wider than about 3 m and/or you cannot see the bottom, picture 3-5 columns, each 0.5 m wide going down to the streambed. Estimate % of water surface and % of water volume occupied by macrophytes within these.

SHMAK assessments do not involve identifying species, e.g. recording invasive species, as this requires some specialist skills.

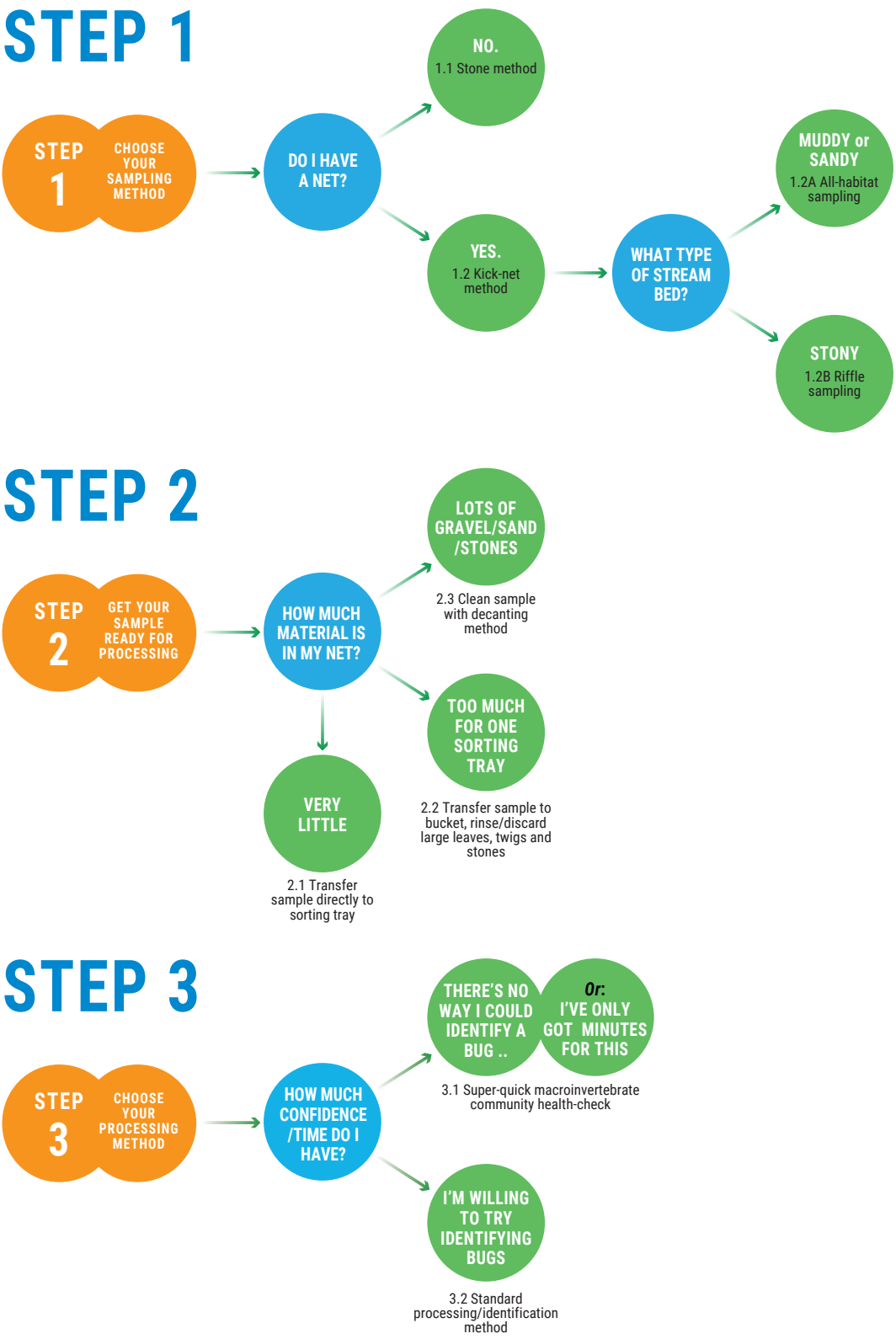
BENTHIC MACROINVERTEBRATES

Benthic macroinvertebrates (also known as "bugs") are typically sampled by the "kick-net" method (*a SHMAK Level 2 method*). In this method the bugs crawling on surfaces (such as streambed sediments, wood or macrophytes) are dislodged by kicking, brushing or jabbing, and collected in a net held just downstream. Samples are normally collected from riffle areas (shallow, fast-flowing areas with a wavy water surface) where the greatest number and variety of species are found. If you don't have a kick-net, you can use the stone method (*a SHMAK Level 1 method*). Although the stone method is quicker and doesn't require additional sampling equipment, you won't collect as many different types of bugs as the kick-net method. This means that if you change from the stone to the kick-net method your results are likely to change.

How often to measure: Minimum of once per year, preferably in late summer. Twice per year (summer and winter) or more gives more data for detecting changes over time, and helps keep you familiar with identifying the different types of bugs. Avoid sampling within 3 weeks of a storm.

Assessment overview:

These are the decisions you will need to make to choose your methods.



1.1 How to use the stone method (Level 1)

Number of people: 1 minimum

2 is quicker, and you can check identifications

Equipment:

Measuring tape

Scrubbing (dish) brush

8 litre bucket

White tray – *draw a 4x3 grid on the bottom with marker pen*

Sieve (mesh 1 mm or less)

Bug box (ice cube tray or similar)

Bug sucker (pipette), eye dropper, tweezers, spoon, or paint brush

Benthic Macroinvertebrate Identification Guide

Instructions

1. Choose a riffle habitat at least 10 m long. Stretch out a string line or measuring tape ("transect") across the stream in two places within the riffle.

   If your stream is <2 m wide: *instead of laying transects across the stream, just collect 10 stones from different parts of the riffle, including the middle and edges of the stream.*

2. Part-fill your white tray with stream water (2-5 cm deep), ensuring you have not scooped any bugs with the water. Put it on flat ground or a picnic table.
3. Select five places on your downstream transect, equally spaced across the stream. At each place, bend down and pick up a stone that is at least 4 cm wide. As you pick it up, slide the sieve underneath it to catch any bugs letting go of the stone. If you are also estimating periphyton cover using the stone method, you can do your periphyton estimate now.
4. Place the stone in your bucket as shown, and move to the next place. Aim to get a variety of stone sizes wider than 4 cm. Once you have five stones in your bucket, take them to your white tray on the bank.
5. Place the stones one by one into the water in the white tray, and swirl them gently in the water to get all the bugs into the tray. To remove the last bugs, wash them off with your wash bottle, pick them off with tweezers or scrub the stone gently with the scrubbing brush. Lastly, tip any bugs remaining in the bucket through the sieve and empty the contents into the white tray.
6. Repeat steps 3-5 on your second (upstream) transect.
7. If you have never identified bugs before and you don't think you can do it, or you only have a few minutes, then try the "Super-quick macroinvertebrate community health check". Otherwise:
8. Identify all the bugs you can see in the tray. If it helps, transfer the bugs to your "bug box" before identifying and counting them.
9. Record each type present on the field data sheet. If you want extra accuracy, you can record the abundance of each type using the categories "present" (1 to 4 individuals present), "common" (5 to 19 individuals present) or "abundant" (20 or more individuals present).

1.2 How to use the kick-net method (Level 2)

Number of people: 1 minimum

2 is quicker and you can check identifications

Equipment:

Kick-net

Scrubbing (dish) brush

Buckets (2)

Disposable gloves

Wash bottle with squirt-type top*

Magnifying glass/hand lens

White tray – 25 cm wide, 5 cm deep - make grid with marker pen, lines spaced 5-10 cm apart

Isopropyl alcohol (optional)*

Bug box (ice cube tray or similar)

Bug sucker/collector (pipette), eye dropper, tweezers, spoon, or paint brush

Benthic Macroinvertebrate Identification Guide

* not supplied with SHMAK

1.2a Instructions: all-habitat sampling in sandy or muddy-bottom streams

1. Choose a length of stream up to 20 m long that includes as many of these habitats as possible: vegetated bank margins, snags and logs, and aquatic vegetation.

   Vegetated bank margins. Consists of bank vegetation trailing in the water, and submerged roots and wood attached to banks.

   Snags and logs. Consists of submerged wood – dead trees, logs, branches and roots.

   Aquatic vegetation (macrophytes). Green leafy plants that are rooted in the stream bed. May be fully submerged or grow up through the water surface.

2. Collect a combined sample from the variety of habitats at your site, in proportion to the amount of each habitat type present (minimum of 1 "jab" per habitat type). Note on the field record sheet how many jabs you took in each habitat, and the proportion of each type of habitat in your stream reach. You will need this information next time, to ensure you collect samples in the same way each time at your site.

3. To sample vegetated bank margins, jab the net vigorously against vegetation and roots along a 1 m length of the bank. Then immediately "sweep" the net through the area against the current 2–3 times with an upward motion to catch the bugs you have dislodged. Do the entire jab motion underwater, but well above the streambed so you don't stir up the bottom mud.

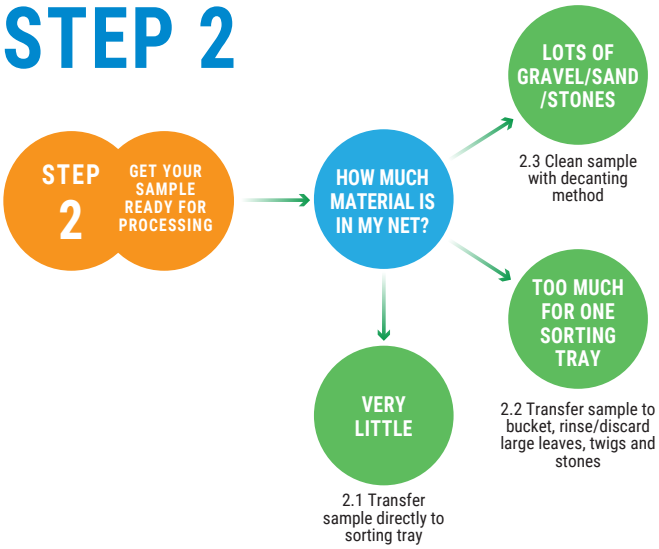
4. To sample branches and small logs, lift each piece out of the water over the net, and pour water over it while gently brushing the surface with a gloved hand (this will need two people). Catch the bugs in the net as they are washed off. Check each branch to ensure you have removed all bugs before placing it back in the stream. To sample snags and logs that can't be lifted, hold the net downstream of the section of the submerged wood with one hand. With the other hand, rub the snag or log, and if the current is slow, sweep the net through the water to catch the bugs you have dislodged. A one metre length of wood equals one jab.

5. To sample aquatic vegetation (macrophytes), jab the net vigorously against or through the submerged plants over a distance of one metre. Then sweep the net 2–3 times through the area you have jabbed (with a slight upward motion) to catch the dislodged bugs. Make sure you keep the net underwater, but well above the streambed so you don't fill it with mud or other debris. If possible, avoid sampling areas with lots of periphyton. Each combination of jabbing and sweeping over a one metre distance equals one jab.

6. Repeat the steps above until you have a total of 10 jabs, with each habitat type sampled in proportion to how abundant it is (minimum of one jab per habitat type).

1.2b Instructions: riffle sampling in stony-bottom streams

1. Choose a riffle habitat at least 10 m long. Riffles are shallow, fast-flowing areas of the stream where the water surface is broken by ripples or small waves.
2. Begin at the downstream end of the riffle. Select a 0.1 m² area of streambed to sample. This is about 30 cm x 30 cm – a square as wide as the opening of your kick-net. The area should have natural flow that will direct animals into the net.
3. Place the net on the streambed. Place one foot into the sampling area immediately upstream of the net and kick the streambed to dislodge the upper layer of cobbles or gravel. Also kick the upper few centimetres of streambed to dislodge burrowing animals. Disturb the sampling area until the area is thoroughly worked over (up to 30 sec, depending on the bed).
4. When finished, use a forward scooping motion to lift the net from the water.
5. Repeat step 3 until you have sampled seven different locations within the 10 m-long study area, each location upstream from the last. Include locations near the centre and sides of the stream, and with a variety of stream flow speeds. This should make a total area of 0.6 – 1.0 m² of riffle habitat.



2. Getting your sample ready for sorting: kick-net (riffle and all-habitat) methods

2.1 If there is not much debris (stones, leaves, etc.) in the net:

1. Invert it and empty the contents directly into the white tray.
2. Gently pick clinging animals off the net and into the tray or bucket using fingers or tweezers, or rinse them off with a few squirts from your wash bottle.

2.2 If the sample has too much debris for one tray:

1. Carefully empty the net contents into a bucket part filled (about 5 cm) with water.
2. Pick or wash clinging animals off the net as described above. If your bucket is nearly full of water after you have washed all the animals off the net, let the debris and animals settle to the bottom of the bucket. Then gently pour the excess water through the centre of the net. If any bugs were caught in the net, wash them back into the bucket.
3. Lift out and rinse off any large debris items (stones, sticks, leaves).
4. If your sample is full of debris, see "Cleaning your sample with the decanting method" (Section 2.3) for how to separate the bugs from the debris.
5. Swirl the contents of the bucket and pour into the white sorting tray as a thin layer, so the bottom of the tray is still visible. If the sample has too much debris for this, you can pour in some at a time. Spread the sample as evenly as you can.

2.3 Cleaning your sample with the decanting method

If the sample in your bucket is full of debris, sand and/or mud:

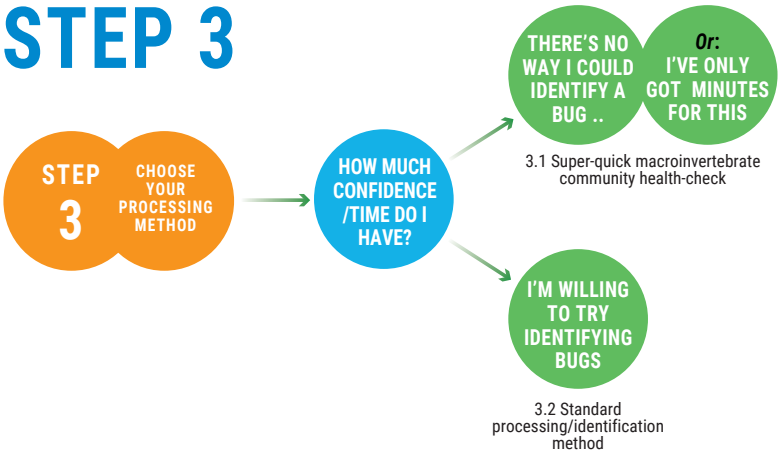
1. Remove large pieces of debris (leaves, twigs, and stones) from the bucket, one piece at a time. Wash any attached animals back into the bucket by swirling each piece in the bucket, or squirting with your wash bottle. Remove any last attached animals using fingers or tweezers and return them to the bucket.
2. Swirl the remaining contents of the bucket to stir up the bugs and light debris (leaves, twigs, etc.) off the bottom then carefully pour the bugs and light debris into the second bucket, leaving the heavier stones and gravel in the bottom of the first bucket.

3. Pour the sample from the second bucket into the net, making sure no bugs are left behind in the bucket.
4. If the sample in the net still contains lots of mud, pour clean water over it to wash the fine mud through the net.
5. Collect some new stream water in the second bucket (ensuring the new water is clean and without bugs) and add it to the first bucket that still has the heavy stones and gravel.
6. Repeat steps 2–4 one to three times more, each time swirling a little more vigorously, until you are sure that no bugs have been left behind with the heavy debris. Check the debris for bugs with heavy shells or cases, e.g. snails or stony-cased caddisflies.
7. Invert the net into the sorting tray and wash the bugs into the sorting tray by swirling the net in the tray and/or using your wash bottle. Remove all the clinging bugs from the net using fingers or tweezers.

Taking your sample home

If you can't (or prefer not to) identify your bugs in the field, or you want to preserve your sample for future reference, you can take your sample home.

1. Once your sample is in the bucket, remove the water by pouring it off through the kick net or sieve.
2. Gently scoop out the sample with your fingers and place in your sample container. Check for any remaining macroninvertebrates in the bucket, on the net or sieve and on your fingers. Pick these into the sample container using fingers or tweezers, or wash them in using your wash bottle.
3. Place a sticky label on the side of the sample container (not the lid) and write the site name, date and sampling method (e.g. riffle or all-habitat) using a pencil. Place a second label with the same information inside the container. This label should be made of waterproof paper.
4. If you intend to keep the sample for more than 48 hours, then add preservative such as isopropyl alcohol (available from hardware stores). Aim for the concentration of preservative in the sample container to be 70–80% (i.e., allowing for the water already present). Be generous with preservative for samples containing plant material (leaves, sticks, macrophyties, or moss).
5. Screw the lid on tightly. Note on your field data sheet the collector's name, sampling method (e.g., kick-net, 0.5 mm mesh) and preservative used.



3. Sorting and identifying

3.1 Super-quick macroinvertebrate community health check

To get macroinvertebrate data for comparing with others you need to identify the bugs using the *Benthic Macroinvertebrate Field Identification Guide*. But if you have never identified bugs before and you don't think you can do it, or you only have five minutes, then try the "Super-quick macroinvertiebrate community health check".

Look into the sorting tray and assess the macroinvertebrate community based on the bugs you can see and the following table:

Most bugs have six legs (though legs may be hidden inside a straight or curved case). Some may look like centipedes. Some have two or three hair-like tails. Some move fast and/or swim.	Health: GOOD
A mix of 'good health' and 'poor health' stream bugs, including some with six or more legs.	Health: FAIR
Most bugs have no legs (may be worm-like or have a shell like a snail or clam) and no hair-like tails. Most move slowly.	Health: POOR

Next time, take the challenge and try identifying the bugs!

3.2 Standard method for sorting and identifying

1. Part-fill each compartment of the bug box with clean stream water.
2. Use suckers, brushes, tweezers or spoons to pick through the debris in the sorting tray, looking for anything that swims, crawls, or is hiding on or in the debris. Look carefully – many of the bugs are quite small and may not move until touched.
3. Work your way from one corner of the sorting tray to the opposite corner using the lines marked on the bottom of the tray so you don't have to look over the same area twice.
4. If you have only a few bugs (e.g. <20) and little debris, you might be able to identify all of them without transferring them to the bug box.
5. Otherwise, sort the bugs into the bug box compartments. Put ones that look similar into the same compartment.

Note: Size and colouration alone are not very useful for distinguishing different types ("taxa") of bugs. While some species are smaller than others, individuals within a species may be different sizes depending on their age.

6. If you have a very large number of bugs, first place one of each abundant type into the bug box. Then look for other types, taking extra care as they may be hidden among the abundant types.
7. Check your bug box to see that each compartment has just one type of bug. Have someone check your sorting before you identify and count your bugs.
8. Identify each type of bug in your bug box, using the Benthic Macroinvertebrate Identification Guide.
9. Use the macroinvertebrate data sheet to record each bug type you have identified.

If you record only which macroinvertebrate types are present, you can calculate the SHMAK Macroinvertenbrate Index (SMI).

If you record the abundance (P for present = 1 to 4 individuals, C for common = 5 to 19 individuals, A for abundant = 20 or more individuals) you can also calculate the SHMAK Macroinvertebrate Abundance Index (SMAI) which you can use to detect small changes in the macroinvertebrate community over time.

Tips for identifying bugs

Photos a great way to confirm, with an expert or teammate, that you identified your bugs correctly, so they are very useful for quality assurance.

- Make sure your photos show the characteristics that help identify each bug (e.g. 3 tails for a mayfly).
- Use a background that provides the right contrast (a white plastic spoon can be ideal).
- Check each photo to confirm that the lighting was good and the important body parts were in focus.

A short video can be useful, as the bug's movement can also help with identification.

Calculating the SHMAK Macroinvertebrate Index – A worked example

5 flat mayflies, 20 smooth caddisflies, 1 green stonefly, 20 midges, 20 mud snails

Tolerance scores are 8, 9, 10, 2, 4

$$\textit{SHMAK Macroinvertebrate index} = \frac{8+9+10+2+4}{5} \times 20 = 132$$

$$\textit{SHMAK Macroinvertebrate Abundance Index} = \frac{(5 \times 8) + (20 \times 9) + (1 \times 10) + (20 + 4)}{5 + 20 + 1 + 20 + 20} = 5.3$$

FISH

Health and Safety: Spot-lighting involves a separate visit to the stream site at night. Working at night involves particular hazards, particularly tripping and falling. Each person in the team should use a torch or headlamp and take special care with placing their feet, especially if entering, exiting or walking through the water. Children should always have an adult buddy.

Getting started: spotlighting is a skill with a lot of small tricks for getting good results. It also involves some special hazards. We recommend working with a professional or experienced person until you are confident.

How to use the spotlighting method

Number of people: 2 minimum

Equipment:

Net (2 nets better) – see Appendix*

Bucket (8 litres or larger) for collecting fish*

Flagging tape*

GPS*

Spotlight or headlamp (30 Watt or equivalent) – one per person if catching fish*

Fish identification guide (see Appendix)

Ruler (can superglue to the bottom of a rectangular tray)*

* not supplied with SHMAK

Setting up a monitoring transect

In daylight, mark a 150 m reach of the stream which includes pools and runs where the calm water offers good visibility (spotlighting is most effective in these habitats). Ensure there are no major tributaries or barriers to fish migration within the reach. Separate it into 10 subsections, each 15 m long. Mark subsections of the reach with flagging tape. Obtain the GPS coordinates for the top and bottom of your reach.

Instructions

1. Begin spotlighting 45 minutes after dark. Record the start time.
2. Walk on the stream bank if possible. Start at the downstream-most subsection of your reach and work in an upstream direction. If you need to stop, stop beside a riffle where the chances of fish moving upstream is reduced.
3. Shine the spotlight 1–2 m ahead, and sweep from bank to bank. Do not scan the beam more than 4 m ahead or you will scare the fish. Identify and count all fish you see (refer to the fish identification guides that you bring with you in the field). If fish are seen but can't be identified record them as "unknown". If you can, capture them and take a photo for later identification.
4. Record how many fish in each fish group you find in your first 15 m subsection. You can record the size of a few fish too, if you like (for this you will need to capture some fish).
5. Move to the next subsection and repeat steps 1-3. Record your finishing time.
6. Each time you spotlight, try to sample the same 150 m reach and try to finish sampling within the same amount of time as the previous sampling (i.e. keep the fishing 'effort' similar for each survey).

Capturing fish

You can capture fish to check your identifications and (if you like) measure a few of each species. Capturing a fish needs two people – the "catcher" and the "assistant". Both have head torches, keeping their hands free for the net.

1. The catcher holds one net downstream of the fish, near the tail end.
2. The assistant holds a second net upstream of the fish, near the head end.
3. Slowly the catcher and the assistant bring their nets together.
4. When the fish enters one of the nets, raise the net rapidly. Do not try to "scoop" or chase the fish with the net.
5. Once the fish is in the net, empty it very carefully into a bucket or clear plastic container containing water. Clear plastic containers will enable you to have a good look at the catch to assist in identification.

Tips for measuring your fish are given in the Appendix.

Tip: Take your time and get your 'eye in'. With patience, most people get good at identifying fish in a relatively short time. Start by identifying fish into the 12 main groups below, and as you gain confidence try to identify individual species. Resources for identifying fish and for finding your nearest Whitebait Connection programme are given in the Appendix.

The following list includes the 12 groups of native and introduced fish (and one crustacean) commonly found in New Zealand streams

Native

- Whitebait and mudfish (Family Galaxiidae)
- Bully (Genus *Gobiomorphus*)
- Eel
- Torrentfish
- Smelt
- Mullet
- Lamprey
- Kōura (freshwater crayfish)

Introduced

- Trout and salmon
- Catfish
- Carp
- Perch
- Live bearers (mainly *Gambusia*)

CURRENT VELOCITY AND STREAMFLOW

How to use the float method

Number of people: 2

Equipment:

Measuring tape

Stopwatch*

Orange (or other item that floats just below water level)*

* means not supplied with SHMAK

Instructions

1. Run the tape measure along a 10-metre length of your reach. Choose a place that is relatively straight, free of obstacles and uniform in width and depth.
2. One person stands at the upstream end, about 2 metres upstream of the measuring tape, holding the orange. The other person holds the stopwatch and follows the orange as it travels downstream.
3. The first person places the orange on the water surface near the middle of the steam and at least two metres upstream of the start of the measuring tape so it is travelling at the speed of current when it reaches the tape.
4. When the orange is in line with the beginning of the tape, the second person starts the stopwatch. Stop the watch when the orange gets to the end of the 10-metre section.
5. Repeat 3 times and average the results.
6. To calculate the current velocity (m/s), divide the distance travelled in metres by the time taken in seconds. Then multiply by a correction factor of 0.86 to compensate for differences in velocity with depth and across the channel (water flows more slowly at the edges than in the middle, and more slowly near the bottom than near the surface).

Current velocity = (distance travelled ÷ average time taken) x correction factor

How to estimate streamflow

We recommend that volunteers discuss with their regional council about ways to use council flow monitoring to estimate streamflow in their stream. However, if there is no flow gauge or hydrometric station at your site for measuring streamflow (or nearby, for estimating streamflow), you can calculate streamflow from the average current velocity in the stream and the cross-sectional area of the water.

Number of people: 1

Equipment:

Measuring tape

1 m measuring stick

Instructions

1. Run a tape measure across your stream within the reach where you calculated current velocity. Measure the "water width" of your stream, i.e. the width of stream that is wet at the time of measurement.
2. Measure the stream depth at 5 to 10 equally spaced points across this cross section. Calculate the average depth from these measurements.
3. Calculate the cross-sectional area (in m^2/s) of this section of stream, multiply your average depth by the stream width.
4. Calculate streamflow (in m^3/s) as cross-sectional area x average current velocity.

STREAM HABITAT

How to do a visual assessment

Number of people: 1 minimum

2 is better: second person can record data and give a second opinion on the estimates, which can make them more consistent.

Marking out your study reach

It's important to assess a reach long enough that your results represent average conditions and are not affected by a single feature such as a tree, an erosion scar, etc. We recommend a length of 50 m, or, if your stream is >3 m wide, a length of 20 times the stream width. If this is too long to be practical, make your reach as long as access, safety and time allow, and try to pick a reach that represents average conditions.

Equipment:

Ruler or measuring stick

You will assess stream habitat based on visual observations of 8 different "parameters" (aspects of the stream habitat), with each parameter accompanied by descriptions on your SHMAK datasheet reflecting a continuum of conditions from excellent to poor.

Some descriptions have two parts, joined by "and". If your site matches both parts of the description, you are in the right category. If it matches only one part, then drop down a category until you are in a category where your stream matches or does better than both parts of the description. In the photo above, large particles (cobbles) cover >75% of the streambed, but only two of the habitat features (boulders and cobbles) are present, so the final score is 4 (patchy and limited). In some descriptions the two parts are joined by "or". If your site matches one part of the description, you are in the right category.

HABITAT FOR AQUATIC ANIMALS	CIRCLE ALL THE HABITAT FEATURES THAT ARE PRESENT:			
	Large wood Root mats Overhanging vegetation Macrophytes Boulders Cobbles			
	Abundant & diverse At least 4 of these habitat features present	Adequate Three of these habitat features present	Patchy & limited Only 2 of these habitat features present	Rare or absent Not more than 1 of these habitat features present
	AND	AND	AND	AND
	Large particles (e.g. cobbles, wood, roots or plants) cover at least 75% of stream bed.	Large particles cover at least 50% of stream bed.	Large particles cover at least 25% of stream bed.	Large particles cover less than 25% of stream bed.
	8 7	6 5	4 3	2 1 0

Score each aspect of stream habitat from 8 (excellent) to 0 (poor), and the stream bank features from 4 to 0. To arrive at each score, first decide which of the four main categories best describes your stream, then decide whether your stream fits better near the top (higher quality) or bottom (lower quality) end of that category.

For each parameter you will need to see the whole of your study reach. At most sites this will involve walking up and down the length of the reach. Read the questions on your SHMAK datasheet.

1. Amount of fine sediment deposited on the streambed

Determine whether your site naturally has a stony bed ("stony-bottom") or naturally has a bed of sand, mud or clay ("soft-bottom").

a) Stony-bottom streams ONLY

Focus on a run habitat. Walk back and forth along the stream bank so you can see the entire run. If you don't have a run habitat, use a riffle.

Look for fine, silty, loose material that looks as if it has settled recently on the stones, wood or water plants that make up the streambed. Recent deposits brush off easily if disturbed. Step into the stream and try the "kick test": gently kick the stream bed or water plants. If this causes the fine material to float into the water, then the deposits are probably recent Estimate what percentage of the stream bed is covered by this loose, silty material.

b) Soft-bottom streams ONLY

Focus on one or more pool areas. Measure the depth of the deepest part of the pool with a long ruler or measuring stick. Then measure the depth of the soft sediments at the bottom of the pool by gently pushing the measuring stick in the sediments until it stops.

2. Habitat for aquatic animals

Count the number of suitable habitat types that are present within your stream reach: large wood, roots, undercut banks, overhanging vegetation, macrophytes, boulders, and cobbles.

3. Flow types

Count the number of different flow types that are present within your stream reach: pools, runs, riffles, chutes, and waterfalls.

For the next three parameters, evaluate the condition of the right and left stream banks separately. Define the "left" and "right" banks by standing at the upstream end of your study stretch and looking downstream. Each bank is evaluated on a scale of 0–4.

4. Bank stability and erosion

Estimate the stability of the banks based on their potential to collapse/erode (e.g. steep banks and those with bare and/or crumbly soil are vulnerable to erosion) and any signs of past erosion. If your stream bank has artificial reinforcing (e.g. concrete, wood or rock lining), then it is probably naturally unstable, so score it low. Score right bank and left bank separately and add the two scores together. Note: left and right are defined as you look downstream.

5. Bank vegetation

Score the main type of vegetation on each bank (up to 10 m away from the stream). If one bank has a mixture of vegetation types, e.g. if there are nearly equal amounts of mown grass and regenerating native vegetation, then chose a score halfway between them.

6. Riparian buffer

Assess the width of riparian vegetation (or the distance to a fence that keeps animals away from the stream) AND how continuous or patchy the riparian vegetation strip is along the stream bank.

7. Riparian shade

Stand in the middle of the stream and look up. Thinking about the path that the sun would travel over a day in summer, how much is the stream shaded by vegetation (leaves, branches, etc.), bank sides, steep valley sides or other objects like buildings? You could think of this proportion as a percentage of the sky over the stream. Zero shading would be in a flat paddock with low banks and no trees or shrubs. Full shading (100%) would be dense vegetation (trees or shrubs) that completely cover over the stream.

Repeat this estimate in at least 3 (better 5-10) different places along your stream reach and write down the average.

8. Channel alteration

Determine how much of your study reach is straightened, deepened or widened, and how severely the flow and channel form are modified by other alterations such as artificial lining of the bank, culverts, or weirs, etc.

STREAMBED COMPOSITION

How to do a visual assessment (Level 1)

How many people: 1 minimum

2 is better (the second person can record the data and also give a second opinion on the estimates, which can make them more consistent).

There are two methods for describing streambed composition: the visual assessment method (SHMAK Level 1 method) is quicker (it should take less than 5 minutes, or you are over-thinking it), while the Wolman walk (SHMAK Level 2) is more accurate.

Instructions

1. Mark out the same area where you collect your benthic macroinvertebrates (bugs).
2. Walking back and forth along the stream bank so you can see your entire study area, estimate by eye the percentage of the stream bed that is covered by each of the particle sizes listed in Table 1. The total of all particle sizes should add to 100%.

Table 1. Size classes of sediment particles.

Particle	Description
Bedrock	Continuous rock
Boulder	>25 cm (basketball sized or bigger)
Large cobble	12-25 cm (grapefruit sized or bigger)
Small cobble	6-12 cm (pool ball sized or bigger)
Large gravel	1.6-6 cm (marble sized or bigger)
Small gravel	2-16 mm
Sand/silt/mud	<2 mm
Man-made (e.g. concrete)	
Large wood	>5 cm diameter
Small wood	<5 cm diameter
Water plants (roots in the stream bed)	

How to do a Wolman walk (Level 2)

How many people: 2

1 to pick up the rocks, 1 to record the data

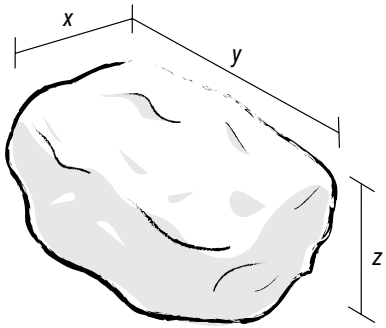
Equipment:

Wolman stick (ruler with particle size classes marked)

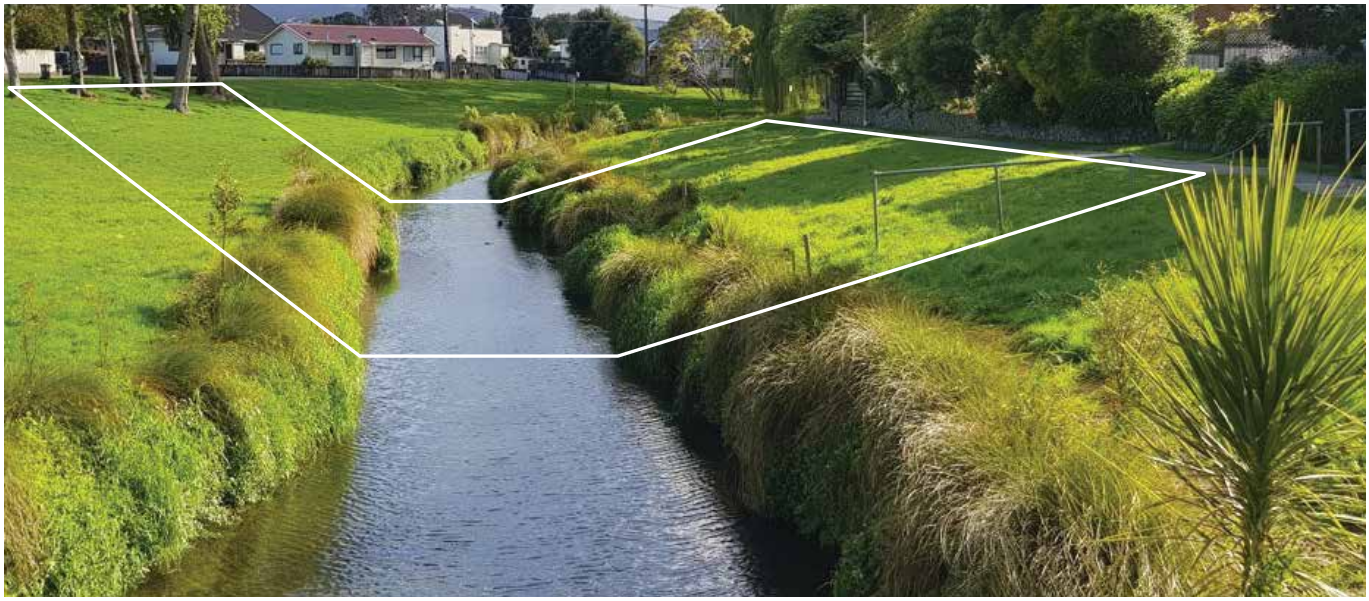
Instructions

1. Mark out the same area where you collect your benthic macroinvertebrates (bugs).
2. Starting at the downstream end of your study area, walk in a zig-zag pattern up the length of the study area. Make sure you reach the side of the stream on each zig-zag. Take large or small steps depending on how big your study area is.
3. At each step or second step, pick up the particle that is immediately in front of your big toe. Try not to look at the particle before you pick it up, as you will be tempted to pick up certain particle sizes more than others.
4. Measure the size of the particle according to the different size classes in Table 1 above, by placing one end of the particle at the end of the Wolman stick and reading the size category that the other end is within (Fig 1). NB: a 3-dimensional particle has three "axes" (x, y, z). You want to measure the second-longest axis (y).
5. For each particle you pick up, put a tick or a "1" in the box beside the appropriate particle size/type in the field data sheet. At the end, count up all the ticks or 1s and calculate the % of particles of each size/type.

Measuring a large cobble.



The three axes of a stone. Measure the *y* axis for the Wolman walk



How to measure the boundaries of your site

RUBBISH

Health and Safety: A rubbish assessment exposes you to certain health and safety risks (e.g., cuts from sharp items, toxic chemicals, pathogens). Wear protective gloves and (if you have one) a pick-up tool for picking up rubbish.

How to do a visual reach assessment

Number of people: 2 minimum

or 3: two rubbish collectors and one recorder

Equipment:

Tape measure (30 m)

GPS (or cell phone with GPS capability)*

Optional items (if you are collecting the rubbish for disposal)

Rubbish bags*

Pick-up claw or kitchen tongs*

Work gloves*

* not supplied with SHMAK

Setting up your monitoring reach

The normal reach length for assessing rubbish is 30 metres, but it may need to be shorter if you can't safely access the entire 30 metres or longer if the stream is large. The width of the monitoring reach (how far away from the stream you look for rubbish) will depend on the characteristics of the site and requires some judgement. Land sloping steeply towards the stream should be included. Decide whether you are collecting rubbish from both banks or only one bank (perhaps because of accessibility issues or time constraints). Record what you decide and do it the same way each time you monitor at your site.

There are two methods for assessing rubbish at your site: the visual reach assessment (SHMAK Level 1 method) gives a quick overview of the amount of rubbish, its likely sources and impacts. The rubbish tally method (SHMAK Level 2 method) provides more detailed data that can be linked to data from other rubbish survey methods (e.g. beach surveys).

Instructions

1. Mark the boundaries of your sampling area, and record the length and width on your datasheet. You may want to use flagging tape to ensure you only assess rubbish within your site boundaries. Record the GPS coordinates of the start and end of your reach or note any landmarks (and take photos) so you can monitor the same area next time.
2. Walk back and forth along the stream bank so you can see your entire reach, looking for rubbish both in the stream and along the stream banks. Look under bushes, logs, and vegetation to see if rubbish has accumulated underneath.
3. Assess rubbish in five categories which reflect the different issues associated with rubbish. Score each category from 8 (excellent) to 0 (very poor). To arrive at each score, first decide which of the four main classes best describes your site, then decide whether your site fits better near the top (higher quality) or bottom (lower quality) end of that class. See page 88.
4. At the end of your assessment, make any notes you feel are important, including potential sources of rubbish such as nearby construction sites or parks. Record whether or not you cleaned up the rubbish and if there is a rubbish bin nearby.

Categories for visual rubbish assessment

- **Amount of rubbish:** This is your first impression of the total amount of rubbish at the site, after observing the entire length of the reach.
- **Threat to Aquatic Life:** Some rubbish items can cause entanglement of aquatic animals (e.g., plastic bags or fishing line). Relatively small and buoyant rubbish items (e.g., plastic pieces) can be mistaken for food items. Toxic chemicals (e.g., batteries, oil or gas cans) can poison aquatic life.
- **Thread to Human Health:** Some rubbish items are dangerous to people who wade or swim in the water (e.g., broken glass, chemical containers). Sites with rubbish that could spread pathogens, such as diapers or pet waste, are rated the worst.
- **Dumping and Littering:** If your site is used for dumping or littering, you will see piles of rubbish, particularly close to a road.
- **Accumulation of Rubbish:** This is the amount of rubbish that has washed down from upstream locations. Accumulated rubbish is distinguished from dumped rubbish by signs that it is old and has been transported by the stream. Faded colours, silt marks, rubbish wrapped around roots, and signs of decay suggest downstream transport.

How to do a rubbish tally

Number of people: 2 minimum

one collector and one recorder. Quicker with more collectors

Equipment:

Tape measure (30 m)

Rubbish bags*

Work gloves*

Pick-up tool claw or kitchen tongs*

Digital luggage scale (optional)*

* not supplied with SHMAK

Rubbish identification

Before you begin your assessment, familiarise yourself with the different types of rubbish which are categorised by material: Plastic; Foamed plastic; Paper, Wood; Glass & Ceramic; Rubber; Cloth; Metal. Items that you can't identify can be written in the "Other" category and described in detail in the Notes section. Taking a photo can help you identify it later.

Instructions

1. Set the boundaries of your sampling area. Record the length and width, and GPS coordinates or landmarks at the start and end.
   - Marking the boundaries with flagging tape can help you see where to assess rubbish.
2. Starting at the downstream end of your reach, walk upstream and collect all the rubbish in the stream, and place in the same bag.
3. Collect and tally all the rubbish on the stream banks (above the high-water mark), including under rocks and vegetation, and place in the same bag.
4. Closely inspect the ground for small items like pieces of plastic or glass. Do not count these items but indicate on the field sheet whether there was none, some (1-25), or many (> 25) pieces.
5. Count and weigh the rubbish in each of the categories (except for hazardous items).
   - Collect all the items in a category into a bag and weigh them using a luggage scale. A kitchen scale can be used for smaller items.
   - Circle the items in each category that weren't weighed (e.g. because they were too heavy. If possible, estimate their individual weight.
   - Count and weigh items found in the stream separate from those found out of the stream.
6. When finished, record on the tally sheet the total number of items in each category found above and below the high-water mark. Complete your data sheet before leaving the site.

APPENDIX

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ADDITIONAL INFORMATION FOR *E. coli* ASSESSMENTS

Incubators

An incubator is necessary to keep your *E. coli* plates between 33 and 37 °C. You can build your own incubator using the instructions below or purchase an incubator.

Build your own incubator

1. You will receive a Styrofoam chilly bin in your SHMAK kit. If it leaks, line it with a plastic liner.
2. Fill the chilly bin with tap water up to about 5 cm from the top. Using slightly warm water (<35 °C) will reduce the time it takes for the temperature to stabilise.
3. Cut a hole in the lid about the diameter of the aquarium heater. Position the hole near one end of the lid so there is enough space inside to float the sandwich box. But make sure the aquarium heater will not touch the side of the chilly bin.
4. Slide the heater through this hole, place the lid on the chilly bin and turn on the heater. Leave it for 6 hours or overnight for the water temperature to stabilise.
5. Before you go to the field, check that the temperature is between 33 and 37 °C. Check again just before you place your sample inside. If the incubator temperature is higher or lower than this, carefully adjust the heater up or down using the purple knob on top, and re-check the incubator after about 2 hours.

Purchase an incubator

Most poultry egg incubators are ideal for growing *E. coli* as they have an adjustable thermostat allowing you to select the appropriate temperature (~35 °C). Poultry egg incubators can be purchased from a poultry or farm supply store or on Trade Me.

Note – some poultry egg incubators incorporate an antimicrobial additive embedded within the plastic itself which prevents the growth of harmful bacteria. Before purchasing an incubator read the product specifications to ensure no additives have been used.

For more information, consult the *Petrifilm Interpretation Guide* https://multimedia.3m.com/mws/media/2362460/petrifilm-ecoli-coliform-interpretation-guide.pdf

E. coli assessment – Petrifilm

The field manual provides instructions for using MCM *E. coli* plates (Ngaio Diagnostics). However, other Select *E. coli* Count (SEC) plates are available. A common SEC plate is the 3M™ Petrifilm™ *E. coli*/ Coliform plate. As some of the steps for using this method differ slightly from the MCM plates outlined in the manual, additional instructions are provided below if you decide to use these plates.

Storage

Once opened, Petrifilm plates should be kept in the freezer and stored in a tightly sealed package to prevent condensation from building up on the plate.

Direct Plate Method

With Petrifilm *E. coli* plates, the bacteria grow on a pink gel containing the growth nutrients (with MCM *E. coli* plates, the growth nutrients are embedded in a white fabric). After adding your sample to the pink gel and rolling the top flap back down, leave the Petrifilm plate undisturbed for at least one minute to allow the gel to absorb the sample.

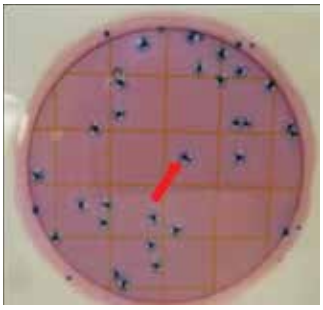
Filtering Method

The Petrifilm plate needs to be pre-wet prior to adding the filter to the plate. Pre-wet the Petrifilm gel using 1 mL of sterile water and a sterile pipette. **Leave the plate for at least 30 minutes to absorb the water.**

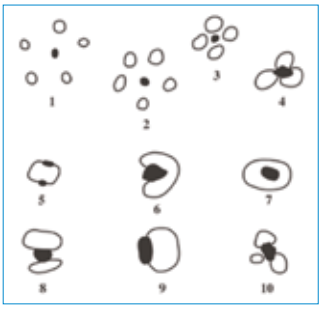
When lifting the clear film to add the filter to the plate, the pink gel will stay attached to the top flap. Place the filter paper on the plate face up. When you roll the film back down, the pink gel will cover the filter paper.

Identifying Colonies

The blue colonies with a gas bubble are confirmed *E. coli* colonies. About 95% of *E. coli* colonies produce gas. The different types of gas bubbles associated with *E. coli* colonies are illustrated in the diagram. You might also see red colonies on the gel. These are other kinds of faecal colonies. Do not count these.



A petrifilm plate is composed of a pink gel in which *E.coli* (blue) colonies and other faecal coliforms (red) can grow.



The different shapes of gas bubbles associated with *E.coli* colonies.

DRAFT

Fish identification and DIY equipment

The NIWA website contains information to show the distribution of all New Zealand's native fish and help predict what fish you might expect in your stream based on their habitat preferences.

http://www.niwa.co.nz/freshwater-and-estuaries/nzffd/NIWA-fish-atlas/fish-finder

http://www.niwa.co.nz/sites/default/files/where_do_fish_want_to_live.pdf

More information on spotlighting protocols can be found in the *New Zealand Freshwater Fish Sampling Protocols, Part 1 – Wadeable Rivers & Streams (Joy, David, Lake; 2013)*

Fish identification

A certain amount of skill and experience is needed to identify the adults of common fish species (e.g. īnanga, banded kōkopu, shortjaw kōkopu, giant kōkopu and kōaro; redfin bully; salmonids), especially if you wish to identify them without catching them. Juveniles are usually harder to identify than adults. To identify them to species you will need to catch them. Alternatively, identify them to genus (e.g. *Galaxias, Anguilla, Gobiomorphus*) and record them as juveniles.

Identification resources are available from the NIWA website and from the Mountains to Sea Conservation Trust's (MTSCT) Whitebait Connection programme (see links below). Some of these identification resources can be printed and taken to the field with you.

http://www.niwa.co.nz/freshwater-and-estuaries/nzffd/NIWA-fish-atlas

https://www.whitebaitconnection.co.nz/teaching-resources/teachers-and-coordinators-resources.html

Equipment

Nets. It is easier to use two nets to capture fish, one behind the fish and one in front to guide it into the net. Your kick net will make a good "guiding net" but you will want a slightly larger net to capture the fish. A long-handled "dip net" is recommended (about 300 mm in diameter, 500 mm deep, and 3-5 mm mesh size). The size of the net can be a little different from these measurements, but the mesh size must be less than 5 mm to ensure that small whitebait don't slip through. Nets can be found at most fishing and hunting stores and even butterfly catching nets will suffice. If you have a net but the mesh size is too large, you can order mesh from Sefar Filter Specialists Ltd. (Auckland) and bring it to a canvas maker.

Fish measuring tray. You need a plain small white tray with a plastic ruler glued in the bottom. You can pick up a plastic tray from any plastics shop and a 30 cm clear ruler at any stationery shop. Superglue is suitable for gluing the ruler into the tray. Measure the total length of the fish to the nearest millimetre from the tip of its snout to the longest part of the tail.

ADDITIONAL SHMAK EQUIPMENT

There are some items not included with the kit that you will need to source yourself. This includes items that you will probably already have at home or can easily purchase from hardware or homeware stores.

Clipboard and pencil	For writing on your data sheets
Bucket 2 L or larger	To collect water for the clarity tube
2 buckets (4 L or larger)	To collect fish and stones for macroinvertebrates (stone method) and to clean debris from macroinvertebrate sample
Rectangular tray with ruler	To measure fish
Batteries	Spare batteries for conductivity meter and phosphate checker
Waratah (Y-post)	To secure temperature logger in streambed
Sledge hammer	To hammer the waratah into the streambed
Wire or cable tie	To secure temperature logger to waratah
Wire cutters	To remove temperature logger from waratah
Mobile phone	For uploading monitoring data, site photos, as a stopwatch, GPS coordinates, downloading temperature logger data
Camera	To take site photos (or use a mobile phone)
GPS	To record location of sampling site or ends of sampling reach
Bottled/distilled water	To dilute your water sample (if nitrate is too high)
Phillips head screw driver	To change the batteries of the phosphate colorimeter
Lint-free cloth or tissue	To clean phosphate vials before inserting in colorimeter
Stopwatch	Necessary for the phosphate test (can use your mobile phone)
Bathyscope	Provides a larger field of view for the periphyton assessment
Marker pen	To mark gridlines on your white tray prior to sorting your macroinvertebrates. Label sample containers and *E. coli* plates
Bottle with squirt-type top	To wash invertebrates off your kick net or sieve
Isopropyl alcohol	To preserve macroinvertebrates if sorting at home
Spotlight	For your fish assessment, 30W bulb is recommended
Dip net	To collect fish (see below)
Fish measuring tray	To measure fish (see below)
Orange	To measure water velocity
Rubbish bags	To remove the rubbish during your rubbish assessment
Pick-up claw tool	To collect rubbish without using your hands
Flagging tape	To mark transects or the boundaries of an assessment area
Aluminium foil	To wrap your *E. coli* water sample and reduce light exposure
Ice packs	To keep your *E. coli* water sample cold
Jar lid	To keep the ziplock bag from pressing on the *E. coli* plate
Bleach	Decontaminate your equipment after use
Kitchen scale	To weigh rubbish
Gumboots/waders	To keep your feet dry and protected in the stream
Warm clothes	Be prepared as weather change quickly
Sun hat and sunscreen	To protect yourself from sun exposure
Water and snacks	Prevent dehydration and keep you motivated
First aid kit	Keep a first aid kit handy at all times
Hand sanitiser	To sanitise your hands until you can wash them
Insect repellent	Sandflies love moving water and you!

GLOSSARY

Accuracy – How close your measurement is to the "true" value. Accuracy can be determined by measuring a sample that has a known value, such as a standard reference sample from a lab, and comparing the measured value to the known (true) value. In the natural environment the true value is not known, so accuracy cannot be assessed.

Aquatic plant – A plant that lives mostly or entirely underwater. Includes periphyton (attached algae) and macrophytes (large aquatic plants, often with roots and leaves).

Aquifer – Underground layers of porous rock or sand through which groundwater flows. This groundwater feeds lakes and rivers. Aquifers are also an important source of well water for towns, cities, farms, and industries.

Base flow – The "normal" flow of a stream, when there is no overland runoff of rain water. Base flow is made up of water that arrives at the stream from shallow and deep underground pathways.

Bias – Where your measurement is consistently higher or lower than the true value.

Benthic macroinvertebrate – Invertebrates are animals with no backbone (e.g. insects, snails, crustaceans). Macro means large enough to be seen by the naked eye. Benthic means living on the bottom of streams and lakes.

Blank – A sample of pure water (e.g. distilled water) that you would expect to give a "zero" measurement, e.g. when testing for dissolved nutrients or bacteria. If it doesn't read zero, you know that something you are doing is contaminating your samples. A "field blank" is added to a sample bottle in the field and picks up any contamination caused by your field and lab methods. A "lab blank" is added to a sample bottle in the lab (or at home). It checks for contamination caused by your lab methods only.

Bug – can mean various things, but in SHMAK it is used as a common name for benthic macroinvertebrates.

Calibrate – adjust a meter (e.g. a conductivity meter) to read the correct value of a known "standard" solution.

Catchment – The area of land from which water flows (by surface or subsurface pathways) into a waterbody. Normally a catchment is bounded by the tops of hills, mountains or ridgelines.

Channel – The physical confines of a stream where water and sediment flow. It is made up of the stream banks and the stream bed. Some channels have water from bank to bank while others have large areas of dry sediment that are only submerged during floods.

Chute – A flow type in a stream where the passage is very narrow (e.g. between two boulders), and water is forced through at higher than normal pressure.

Current velocity – The speed that water travels along a stream channel.

E. coli (or *Escherichia coli*) – A bacterium commonly found in the intestines of warm-blooded animals (mammals and birds). It is used as an indicator of the presence of faecal pollution in freshwater.

Faecal pollution – Contamination of water with animal faeces and potentially disease-causing pathogens.

Field replicates – Two or more measurements or samples collected and analysed from the same site. Replicates can be used to check for either precision or representativeness in sampling.

Flow conditions (also known as State of flow) – How high the streamflow of your stream is today compared to normal (base flow).

Freshwater management unit – A catchment or group of catchments defined by a council to be managed together as a unit under the National Policy Statement for Freshwater Management.

Hard-bottom – See stony-bottom

Impervious – Surfaces such as rooftops, paved areas, and compacted soils where water runs off instead of filtering through. Impervious surfaces increase the rate that water and pollutants in it are delivered to streams.

Incubator – Equipment that creates a warm environment so bacteria added to an *E. coli* plate can multiply and form visible "dots" on the plate. A water bath incubator can be made by filling a chilly bin with water and inserting an aquarium heater.

Indicator – A measurable characteristic or property of fresh water. Includes physical, chemical and biological properties.

Left bank (or true left bank) – The bank of a stream on your left when you are facing downstream.

Macrophyte – An aquatic plant that grows in or near the water and is large enough to be seen by the naked eye. Macrophytes can be either emergent (rooted in the stream bed but growing out of the water), submergent (entirely underwater) or floating (leaves float on the water surface).

Microbe – see microorganism

Microcoleus (previously known as Phormidium) – A type of cyanobacteria (often called "toxic algae") that forms dark brown or black mats on river beds. It can produce toxins that have killed dogs and may cause a health risk to humans swimming (though this is not confirmed).

Microorganism – An organism that can only be seen with the help of a microscope. Often (but not always) single-celled. Common examples are viruses, bacteria, fungi, algae and protozoa.

Order – 1) a way of describing stream size (see Stream order); or 2) a group of organisms, for example mayflies, stoneflies and caddisflies are three common orders of stream insect.

Organism – A living thing, i.e. a plant, animal or microbe.

Parameter – See "indicator".

Pathogen – A bacterium, virus or other microorganism that causes disease.

Periphyton – The community of microbes (algae, cyanobacteria, bacteria and fungi) that are found attached to the surface of submerged stones, wood or aquatic vegetation. It is commonly used to mean only the algae and cyanobacteria.

Pool – Deep areas of the stream where the water flows very slowly and the water surface is smooth. The streambed in pools is often covered by deposits of fine sediment.

Precision – How similar repeated measurements are when collected by the same person. Repeated samples are often called replicates. Precision does not tell you how accurate a measurement is because you don't know the true value.

Quality assurance (QA): The overall plan, including study design, monitoring protocols, training, quality control, and data management, that promotes data quality. QA begins well before you step in the stream.

Quality control (QC): The steps that are in place to control error while you are conducting your monitoring (e.g., collecting replicates, checking field instruments). These steps ensure the monitoring results are representative of the overall condition of the sample area. QC procedures can be internal (done by members of your group) or external (done by outside professionals).

Reach – A length or section of stream that you define for a particular purpose, e.g. for monitoring particular indicators.

Reagent – A chemical or mixture of chemicals added to a water sample to test for a particular substance in the water. The reagent undergoes a chemical reaction with the substance in the water that is being tested.

Replicates – two or more measurements or samples collected and analysed from the same site. Replicates can be used to check for either precision or representativeness in sampling.

Representativeness – How well a measurement represents the overall condition of the stream. It is typically affected by where you sample in your stream. For example, water quality measurements are more representative when they are taken in the main flow of the stream.

Reproducibility – Where two different people or agencies, working independently, get similar results. Reproducibility implies (but does not quite prove) accuracy. Where accuracy cannot be assessed (e.g. most field situations), reproducibility is the next best thing.

Resolution – The ability of a method or instrument to show different levels of an indicator (e.g. small differences in temperature).

Riffle – A shallow, fast-flowing area of the stream where the water surface is broken by ripples or small waves as the water flows over a rough bed.

Right bank (or true right bank) – The bank of a stream on your right when you are facing downstream.

Riparian zone – The margin of a stream (or lake) that includes the banks and the land up to about 20 m from the water's edge. Riparian zones represent the transition between land and water habitats. They are often rich in biodiversity and have a particularly strong influence on the stream or lake. They are often regarded as part of the aquatic ecosystem, although they are above water.

Run – A reach of a stream where the water is obviously flowing but the water surface is smooth or nearly smooth. Runs are usually deeper than riffles.

Sensitivity – The ability of a method or an instrument to detect very low values.

Soft-bottom – a streambed made up mainly of sand or mud.

Species – A group of individuals having common characteristics and capable of reproducing with one another. For example, common bully and upland bully are different species.

Standard solution – A solution containing a precisely known concentration of a chemical or a substance. Standard solutions are often used to calibrate an instrument (e.g. a conductivity meter) or check the accuracy of a chemical test.

State of flow (also known as flow conditions) – How high the streamflow of your stream is today compared to normal (base flow).

Stony-bottom – a streambed made up mainly of large stones, i.e. gravels, cobbles, boulders and/or bedrock

Stormwater – Rainwater that runs off the land (usually paved or compacted surfaces in urban or suburban areas) and often routed into drainage systems in order to prevent flooding.

Stream order – A measure of stream size that is defined by where in the stream network the stream is located. First order streams occur at the top of a catchment and have no tributaries. Where two first order streams come together they form a second order stream. Where two second order streams come together they form a third order stream, and so on. However, when a second order stream is joined by a first-order stream, it remains a second order stream. In New Zealand the largest rivers are eighth-order.

Streamflow – The volume of water per unit time flowing past a point in the stream.

Sub-catchment – If a catchment is defined as the land area that drains to a particular stream, a sub-catchment is the land area that drains to one of its tributaries.

Taxon (plural taxa) – A group of related organisms that are classified together, i.e. they are thought of as a unit. "Low" taxa are grouped together to form higher taxa. For example, several species can be grouped within a genus, several genera can be grouped within a family, several families can be grouped within an order. A taxon could be an informal grouping that is not strictly scientific, e.g. "flat mayflies" in SHMAK.

Tributary – A stream that flows into a larger stream or lake. A tributary does not flow directly into the ocean.

Turbidity – A measure of the relative clarity of water. Water with high turbidity is cloudy (or turbid) while water with low turbidity is clear. Typically measured in nephelometric turbidity units (NTU).

Variable – See indicator.

Wadeable – A stream that is shallow enough to be waded across safely at normal flows. This is normally about knee-deep at moderate current speed, but could be waist-deep in very slow flow.

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