CHAPTER 4
FIELD MANUAL

CONTENTS

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).

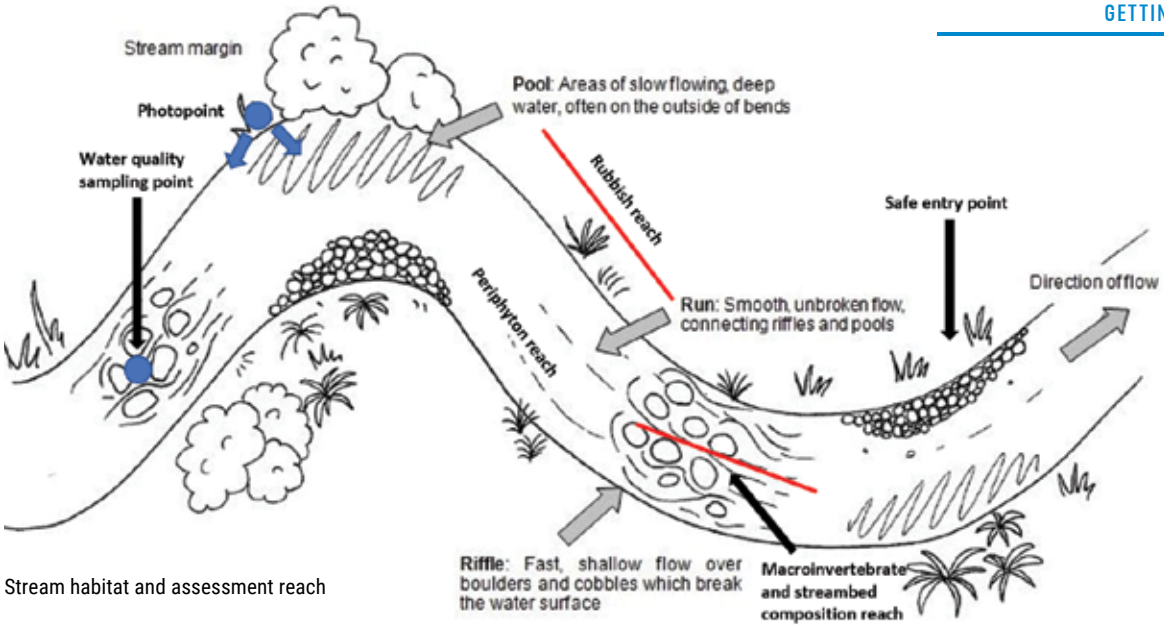
A RIFFLE is a shallow, fast-flowing area of the stream where the water surface is broken by ripples or small waves as the water flows over a rough bed.

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.

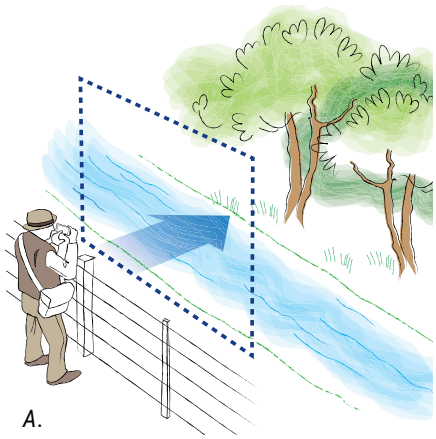
Site information includes:

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

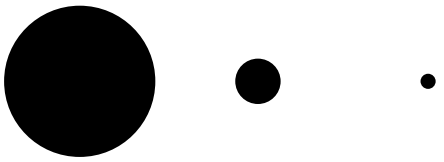
Measuring tape

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.

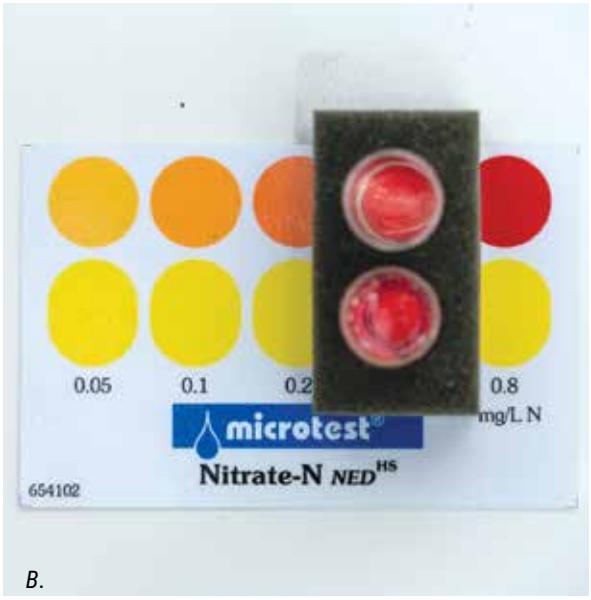
1. Clean the tip of your meter by washing in tap water and wiping with a tissue.
2. Dip the meter into the conductivity standard solution in your kit and note the reading. If it doesn't match the label on the bottle you will need to recalibrate your meter.
3. Adjust the meter so it shows the correct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

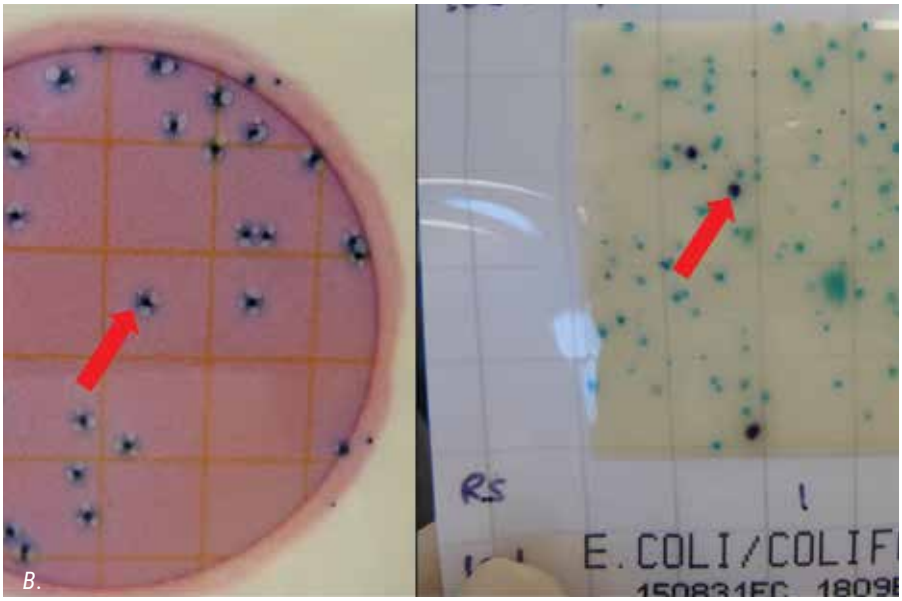

A.

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.

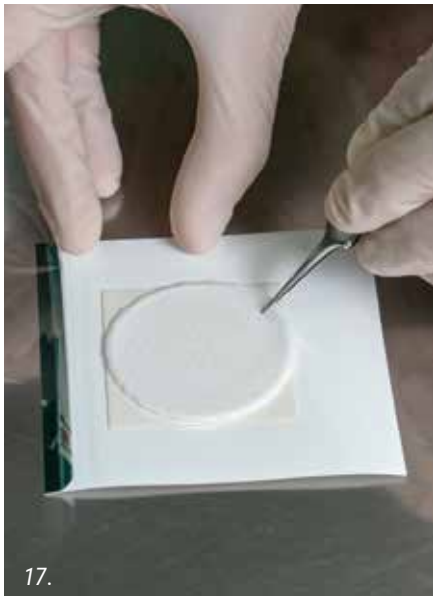
How many plates should I prepare?

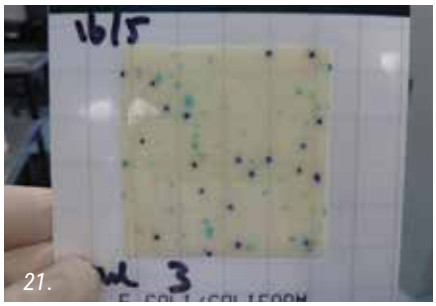
You will get the best results if you end up with 20-80 colonies on the plate. If you expect your site has high levels of *E. coli* (>500 cfu per 100 mL) then the **Direct Plating** method will give the best results. If you think your site has fairly low numbers of *E. coli* (<500 cfu per 100mL) and the 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.

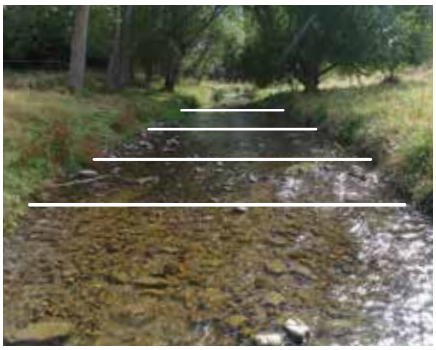
Where to do your periphyton assessment?

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

TOP

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

Caption B. Create a transect along a run section.

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.

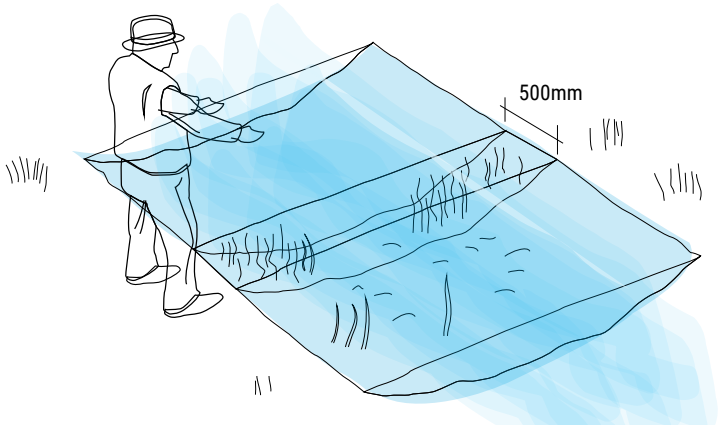


Fig. 1 If the stream is narrow and the water is clear, picture a 0.5 m wide band across the stream. Estimate % of water surface and % of water volume occupied by macrophytes within this.

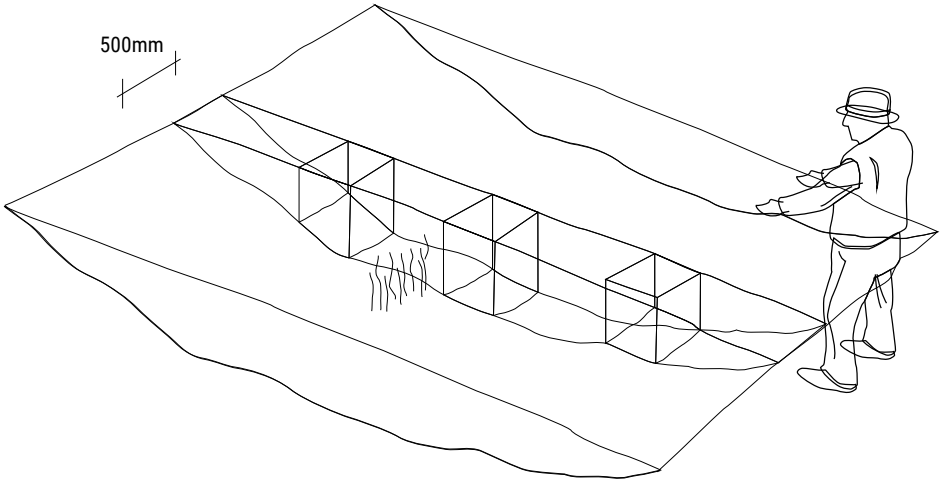


Fig. 2 If the stream is wider than about 3 m and/or you cannot see the bottom, picture 3-5 columns, each 0.5 m wide going down to the streambed. Estimate % of water surface and % of water volume occupied by macrophytes within these.

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

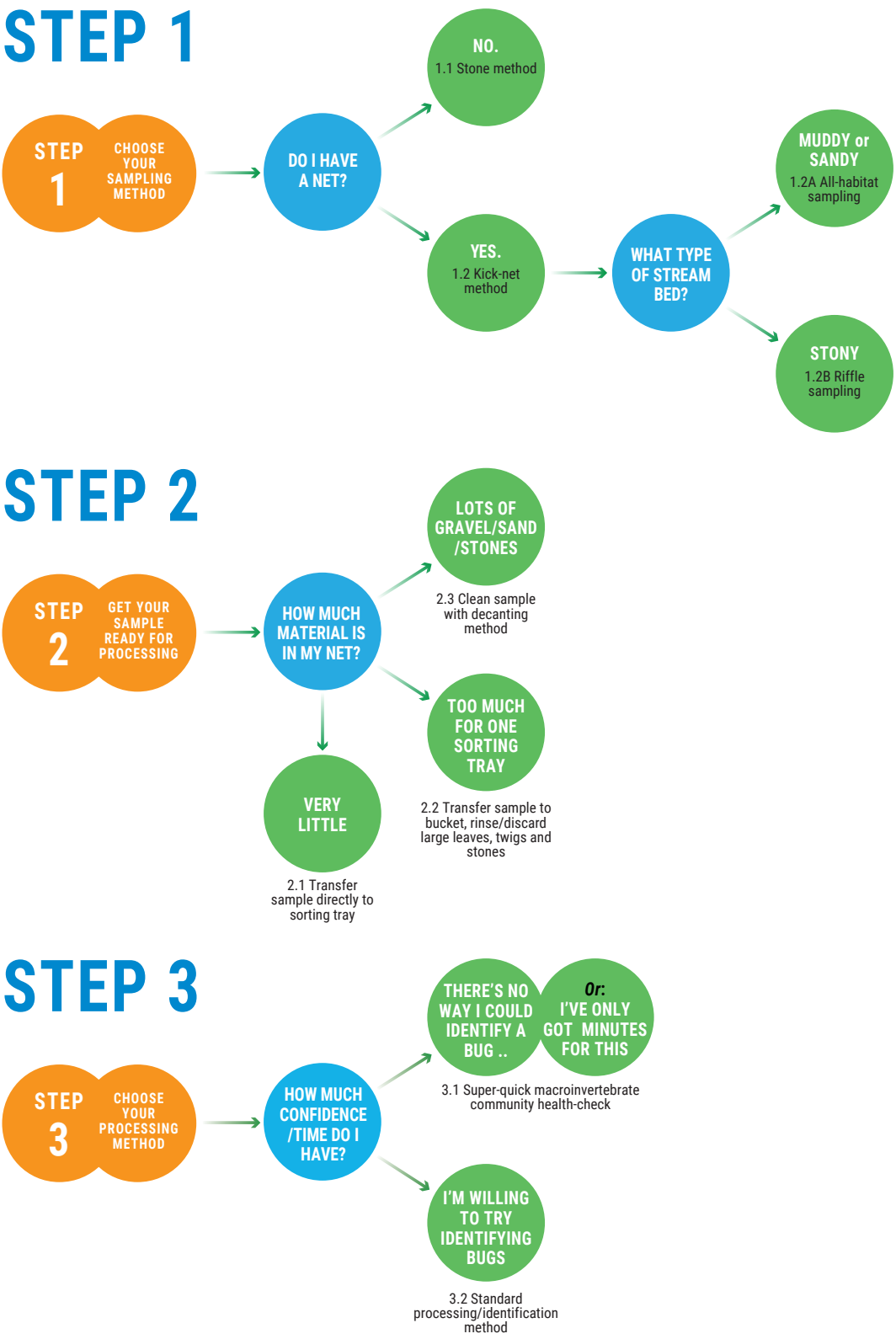
BENTHIC MACROINVERTEBRATES

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.

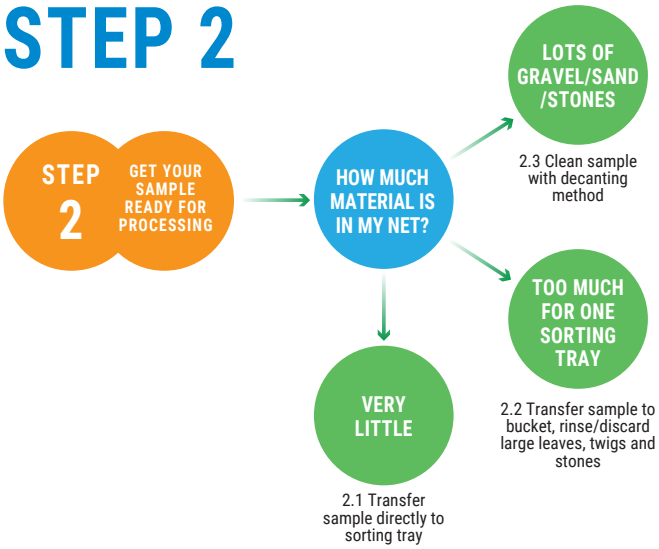
4. To sample branches and small logs, lift each piece out of the water over the net, and pour water over it while gently brushing the surface with a gloved hand (this will need two people). Catch the bugs in the net as they are washed off. Check each branch to ensure you have removed all bugs before 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:

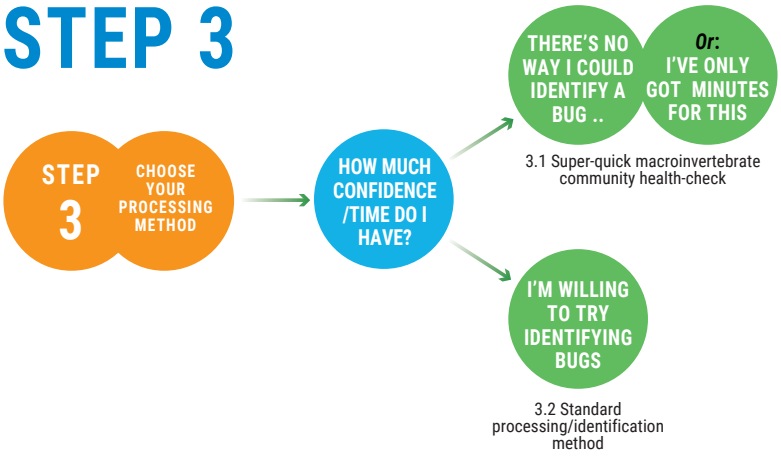
1. Remove large pieces of debris (leaves, twigs, and stones) from the bucket, one piece at a time. Wash any attached animals back into the bucket by swirling each piece in the bucket, or squirting with your wash bottle. Remove any last attached animals using fingers or tweezers and return them to the 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

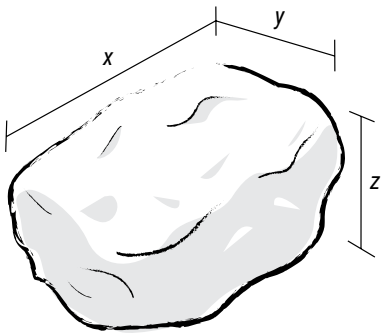
Equipment:

Wolman stick (ruler with particle size classes marked)

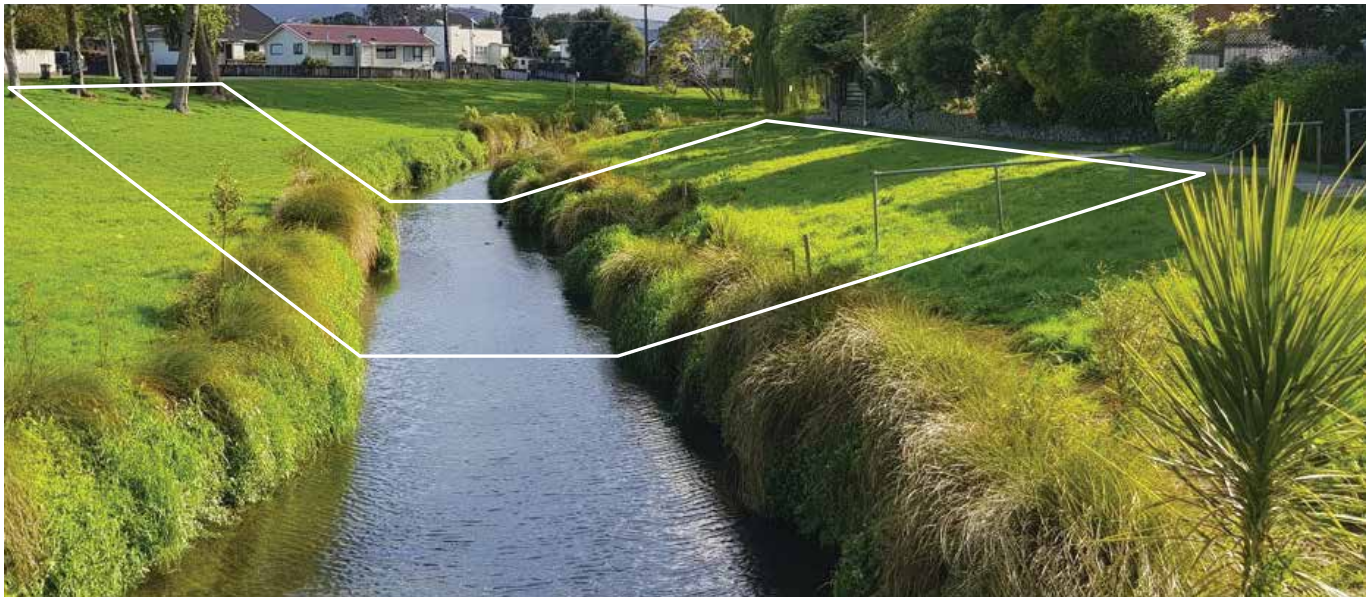
Instructions

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.