


Whitebait Connection (WBC) freshwater investigation monitoring factsheets

 Use this laminated flipchart after the catchment assessment, to prepare and deliver the in stream monitoring experience and to record results while on that freshwater investigation.

 Take lots of photos during the experience, for creating a follow up photo story.



Whitebait Connection Waterway Investigation Safety Briefing Checklist



To carry out before waterway investigation:

- Site pre visit – identify hazards and best entry/exit points
- Consult and notify landowners
- Weather check – keep in minds that rivers and streams may be in flood in the days following rain
- Ratio check – ensure you will have enough adults on the day for a 1:4 ratio (depending on ages)
- Ensure a RAMS form has been completed and sent to the leader of your group.

Items to carry whenever carrying out a waterway investigation:

- Hand Sanitiser
- Sun block
- First Aid Kit
- Fully charged mobile phone
- Rope to throw to people – especially in fast flowing streams and rivers
- Cones to clearly mark boundaries
- A whistle (to be carried by WBC leader at all times)
- Completed RAMS and laminated emergency procedure

Once onsite (before briefing starts):

- Carry out a head count – including adults
- Clearly mark the boundaries and identify the hazards



BETWEEN WATERWAYS

On site safety briefing:

Welcome and objectives

- ❑ Welcome to everyone – introduce yourself and any other volunteers/staff. Introduce the local catchment area including name of waterway – invite karakia and any local stories about the area if appropriate.
- ❑ Housekeeping – location of nearest toilets, first aid, hand sanitiser, resources, any hazards etc.
- ❑ Introduce stream investigation kaupapa – look and learn only. No harming stream life, their habitats, or the surrounding environment. Leave the area in a better state than you found it – not worse! Be kaitiaki. Ensure everyone has clean shoes.
- ❑ Go over the objectives and the students's inquiry questions e.g. learn about local waterway, identify freshwater animals, learn how to tell if a waterway is healthy, learn how to help a waterway. Ask permission to take photos.

Health and safety

- ❑ Identify hazards for the group to be aware of as you see fit – e.g. flowing water, warmth, contaminants in the water, slippery surfaces, getting lost, sun/wind/rain, any site specific hazards, do not drink the water, Take care on rocks
- ❑ Identify shoes policy – if sharp objects present students must wear shoes, if deemed safe students are to take care when shoes not on
- ❑ Identify boundaries
- ❑ No students are to go into the water above the knee
- ❑ Whistle – 1 blow on whistle means to stop and listen, 3 blows on whistle means to immediately gather group and meet at pre-determined emergency meeting point.

What to look for – using ID pictures

- ❑ Ask them what they think they will find and then outline what they may see e.g. fish, Eels, Koura, macroinvertebrates, plants.

Logistics and Biosecurity Briefing

- ❑ Check, clean & dry – importance on participants not transferring invasive plants & animals between waterways
- ❑ Equipment briefing – including how to care for equipment – more details later.
- ❑ Student grouping – ensure all student know which group they belong to and their adult. No students to go outside their groups designated area, or leave view of their adult.



BETWEEN WATERWAYS

Checking the fish traps



Please handle with care! Wet your hands and try not to handle them directly if possible



Identify, measure, photograph, record and release





INANGA
GALAXIAS MACULATUS
80 - 100 mm



KOARO
GALAXIAS BREVIPINNIS
160 - 180 mm



SHORTJAW KOKOPU
GALAXIAS POSTVECTIS
150 - 200 mm



BANDED KOKOPU
GALAXIAS FASCIATUS
200 - 260 mm



GIANT KOKOPU
GALAXIAS ARGENTEUS
300 - 400 mm



*** EEL/TŪNA**
ANGUILLIDAE
Male 700mm, Female 2000mm



TORRENT FISH
CHEIMARRICHTHYS FOSTERI
100 - 120 mm



LAMPREY
GEOTRIA AUSTRALIS
450-750 mm



COMMON BULLY
GOMIOMORPHUS COTIDIANUS
100 mm (males larger)



CRAN'S BULLY
GOMIOMORPHUS BASALIS
80 mm (males larger)



BLUE GILLED BULLY
GOMIOMORPHUS HUBBSI
50 - 60 mm (males larger)



REDFIN BULLY
GOMIOMORPHUS HUTTONI
80 - 120 mm (males larger)

- GALAXID
- BULLY
- OTHER SPECIES

*** LONGFIN EELS:**
DORSAL FIN LONGER THAN VENTRAL FIN, DOMED ADULT HEAD, THICK RUBBERY LIPS, SKIN CREASES, FEMALES 2 M LONG, DARK BROWN TOP, GREYISH YELLOW BELLY

***SHORTFIN EEL"**
SMALL TAPERED HEAD, SKINNY LIPS, DORSAL AND VENTRAL FINS SAME LENGTH, SMOOTH SKIN, FEMALES 1 M LONG, OLIVE GREENTOP, SILVER/WHITE BELLY

Assessing barriers to fish migration

A bit of background: An unusually high number of our native fish are diadromous. This means that they migrate between fresh and salt water as a necessary part of their lifecycle.

- o Species such as eels, mullet, freshwater flounder grow to adulthood in freshwater and then migrate downriver to breed in the sea.
- o Galaxiids, smelt and bullies breed in freshwater with their juveniles travelling downriver to develop at sea.
- o Only the lamprey breeds and develops in freshwater streams and spends its entire adult life at sea.

Because these diadromous species all need to migrate upriver from the sea at some stage, they are vulnerable to barriers created by falls, chutes, dams, weirs, and perched culverts. This is one of the most significant causes of the decline in freshwater fish populations in New Zealand.

Test it: If climbing species are absent from your site, there is likely to be a barrier to the upstream migration of the juveniles downstream. Check your site's altitude and distance upriver from the sea using the information you have gathered about your catchment. Have you already identified any barriers to fish passage in these initial investigations? You can carry out some observations on site by looking around and noting down any partial or total barriers you find (some examples are shown in the pictures over the page...



Partial barrier - swimming species unable to pass



























Partial barrier - swimming species unable to pass



Total barrier - swimming and climbing species unable to pass



Total barrier - swimming and climbing species unable to pass

Stage	Name of test
<p>One - Physical habitat parameters and first Impressions</p> 	<ul style="list-style-type: none">  Date, time, group name, location, name of waterway. Weather and recent rainfall and GPS coordinates  First Impressions- observation inspired conclusions 'a feeling in your puku'  Optional cross-section of stream and sketch of stream and catchment area  Watercourse Type and Catchment Landuses  Mesohabitats  Channel Type and Lining  Floodplain connectivity  Substrate embeddedness and particle size  Organic Matter Abundance  Riparian Vegetation and Shade
<p>Two - Water Quality (Non Biological Tests)</p> 	<ul style="list-style-type: none">  Clarity  Conductivity  Temperature  Flow  pH  Nutrients - phosphates and nitrates
<p>Three - Testing the Life (instream biota)</p>	<ul style="list-style-type: none">  Water sample option - bacteria  Choosing a site  Preparing the equipment  Sampling the different microhabitats - Kick Sampling, Scoop Netting, Picking up rocks  Identification and sensitivity scores  Kaupapa - how to take care of your findings

Stage One

Physical habitat parameters and first impressions



Date: / /

Time:

Group Name:

Location:

Weather: Sunny Cloudy Raining

Recent Rainfall: None Light Moderate Heavy
(within last two days)

GPS Coordinates:
(optional)

First Impressions

Note down here any observation inspired conclusions or “feelings in the puku” about the stream health...ask participants to rate the waterway out of ten (1 being dead and 10 being very healthy and not in need of any assistance).

Sketch of catchment and stream

Include key land uses and landmarks

Using the WaiCare Invertebrate Field Guide (back pages) choose from the following categories:

Watercourse Type: Stream, Wetland/Pond, Estuarine

Known Catchment Uses: Bush, Pasture, Urban

Vegetation/Shade: Shady, Partly Shaded, Unshaded

Bank Stability: Stable, Some erosion, Very unstable

General Waterway Substrate/s: Muddy, sandy, weedy, gravelly, stony, concrete

Instream microhabitat/s: mud scrape, open water, macrophytes, root systems, woody debris, stones

Mesohabitats

Rapid - shallow to moderate depth, swift flow and strong currents, surface broken with white water

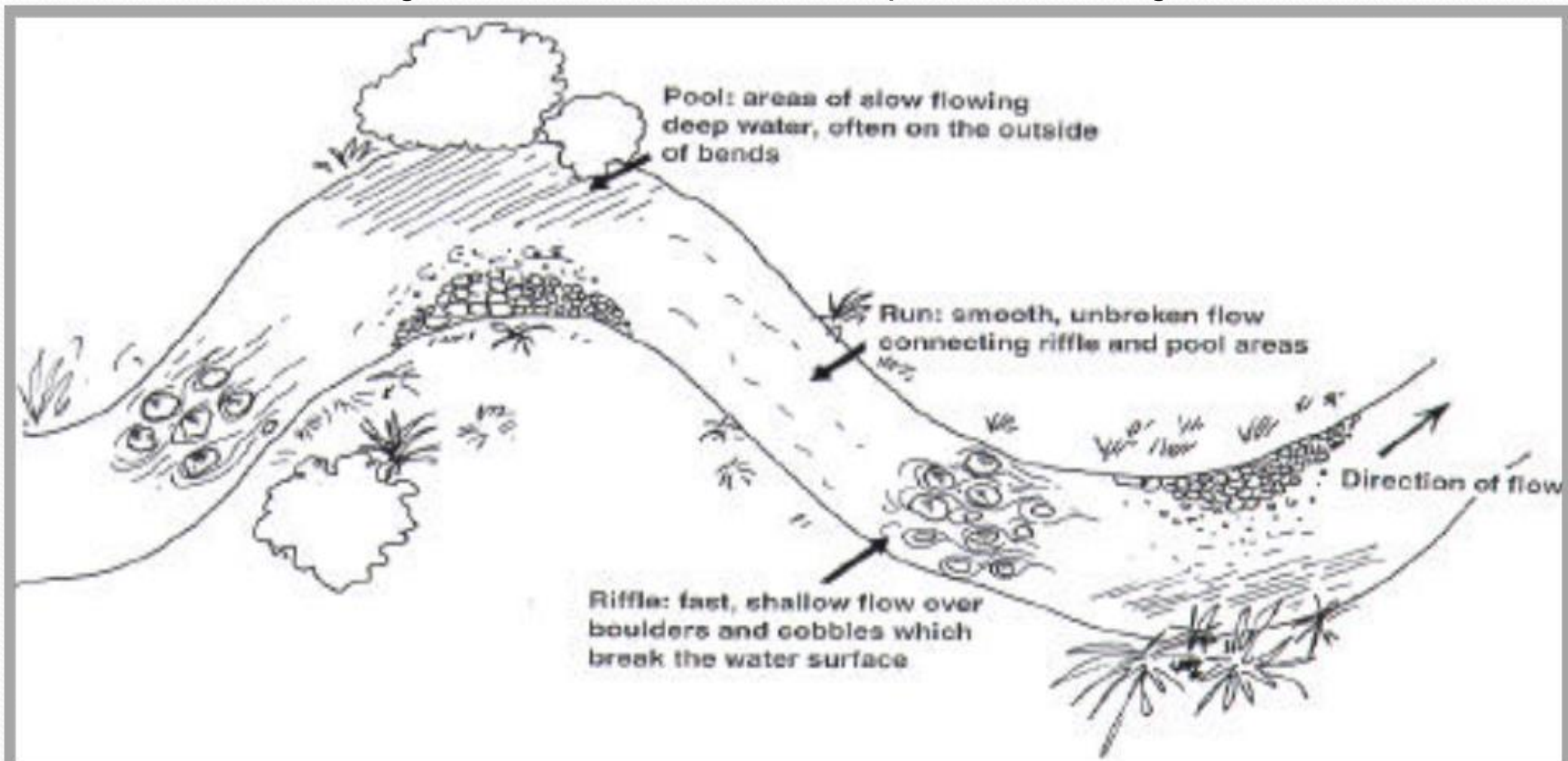
Pool

Riffle

Run

(All these are described below...)

Backwater - slow or no flow zone away from the main flowing channel that is a surface flow dead-end; although flow could downwell or upwell from the groundwater zone.



Channel width and depth

A bit of background: The wetted channel width provides a measure of the available habitat to stream life. Studies have shown that channel width narrows with the removal of riparian vegetation and conversion to pasture. Equally, when fenced and restored, the channel widens out again to the original. This is attributed to the replacement of pasture grasses (that hold the banks) with woody riparian plants that allow erosion during storm events. A wider channel provides more habitat for stream life. Water depth is also an important factor of the stream habitat availability as it influences mesohabitats.

Measure it: A tape measure can be used to measure water width, perpendicular to stream flow at, at least 10, evenly spaced transects along your stream study reach. The reach surveyed should ideally be at least 20 times the average channel width with a minimum reach length of 50 metres. Measure water depths at each transect of the 10 transects at 10%, 30%, 50%, 70% and 90% of the distance across the channel (i.e. make 5 depth measurements at each cross section). This will give you an average depth and, if plotted, these figures will give you approximate cross sections of the stream channel at each transect.

Natural Flow regime

A bit of background: The natural flow regime of a river or stream is the normal cycle of high, normal and low flows that a watercourse might experience within an unmodified catchment. Changes to the natural flow regime in a stream reach can change its ecological character in several ways.


For example, an increase in water velocity due to an input of water from a stormwater pipe or increased stormwater run-off from a more developed catchment, may result in greater erosion of a stream channel and flood scour of habitats and macroinvertebrates. Conversely, retention of water by a dam may affect stream morphology through changes in sediment deposition or the natural run-riffle-pool structure. Straightening a stream and a concrete lining may help reduce floods but loss of meander pattern and natural substrate significantly reduces habitat opportunities.

Measure it: Search your stream reach for stormwater inlet pipes. How many are there? What size are they? The more pipes and the greater their size, the more effect there will be on flow levels.

Similarly search for dams or places where there may be a water take from your stream reach.

In your stream reach try to determine the extent of channel modification. Tick which of the categories below your stream best fits into over the page.

Channel Type...

Channel type	Tick box that best suits your stream	Naturalness of flow regime
Natural channel with no modification		<div style="text-align: center;"> <p>High</p>  <p>Low</p> </div>
Natural channel, but flow patterns affected by a reduction in roughness elements (e.g. woody debris, or boulders)		
Channel not straightened or deepened, but upper banks widened to increase flood flow capacity		
Natural channel, but evidence of channel incision from flood flows		
Natural channel, but flow patterns affected by increase in roughness elements (e.g. excessive macrophyte growth)		
Flow patterns affected by artificial in-stream structure (e.g. ponding due to culvert, weir or unnatural debris)		
Channel straightened and/or deepened		

NB: Use photographic examples from the SEV Pages 31-33...as shown over page



Natural channel with reduction in roughness elements



Upper bank widened to increase capacity



Natural channel shape, but evidence of channel incision (note the flood debris in the tree is below bank full height)



Channel affected by increase in roughness elements




Flow patterns affected by artificial instream structure



Channel straightened and deepened

Type of Channel Lining

Type of channel lining	Tick box that best suits your stream	Naturalness of flow regime
Natural channel with no modification		<div style="text-align: center;"> <p>High</p>  <p>Low</p> </div>
Bed with unnatural loading of fine sediment		
Bank OR bed lined with permeable artificial lining (e.g. gabion baskets)		
Bank OR bed lined with impermeable artificial lining (e.g. concrete)		
Bank AND bed lined with permeable artificial lining		
Bank AND bed lined with impermeable artificial lining		

NB: Use photographic examples from the SEV Pages 35-37...as shown over page



Natural channel with no modification



Bed with unnatural loading of fine sediment



Bed lined with permeable artificial lining



Banks lined with impermeable artificial lining




Bank and bed lined with permeable artificial lining



Bank and bed lined with impermeable artificial lining

Floodplain connectivity

Floodplain description	Tick box that best suits your stream	Connectivity to floodplain
<p>Movement of flood flows onto and across the floodplain is not restricted by any artificial structures or modifications.</p>		<p style="text-align: center;">High</p>  <p style="text-align: center;">Low</p>
<p>Floodplain present, but connectivity to the full floodplain is restricted by modification (e.g. stop banks or urban development).</p>		
<p>Floodplain present, but connectivity to the floodplain reduced by channel incision or bank widening so that most flood flows are unlikely to reach the floodplain.</p>		
<p>No hydrological connectivity with the floodplain as all flows are likely to be artificially contained within the channel.</p>		

NB: Use photographic examples from the SEV Page 39...as shown over page



Floodplain connectivity not restricted by artificial structures



Floodplain connectivity restricted by development



Floodplain connectivity restricted by channel incision



Floodplain connectivity restricted by modified channel

Substrate Embeddedness

A bit of background: This is the extent to which rocks (gravel, cobble, and boulders) are buried or surrounded by fine sediment. Rocks may become smothered when large-scale sediment inputs occur upstream. Reduced stream flow may in turn speed up deposition. As rocks become embedded there is less space between and under rocks for colonisation and the stream community will become dominated by groups such as worms and midges.

Measure it: Look for how much of the streambed is buried by silt or fine sediment. Observations of embeddedness should be taken in the middle of rocky/cobbled areas. Reaching into the stream and unplugging a few rocks is a good way to estimate the covered depth if it is difficult to estimate by sight alone.

Estimated depth of substrate embeddedness =

Substrate Particle Size

1. Lay tape measures across your stream reach at 6 positions including 2 riffles, 2 runs, and 2 pools.
2. At each cross section, randomly select 10 particles while wading across the stream. To achieve random selection, pick up the particle immediately in front of your boot at each step across the stream. If the particles are completely covered in a layer of fine sediment (i.e. the first particle touched is sediment and not the larger particle beneath), and if you are able to pick the sediment up without pinching finger tips together (to avoid overemphasising transient fine deposits of silt/sand), then record that particle as silt or sand.
3. Measure the length of each particle's second-longest axis using a "Wolman stick" or ruler and then assign it to one of the categories in the table below.

Size category	Category name	Size category	Category name
<0.063mm	Silt, mud	16-64mm	Large gravel
0.063-2mm	Sand	64-128mm	Small cobble
2-4mm	Small gravel	128-256 mm	Large cobble
4-8mm	Small medium gravel	256-4000mm	Boulder
8-16mm	Medium large gravel	>4000mm	Bedrock

Organic Matter Abundance

A bit of background: The input of organic matter (e.g. leaves, wood) into the stream from external sources, coupled with in-stream production by plants and algae via photosynthesis, is an important driver of biological production in a watercourse. The amount of organic matter on the stream bed is a good indicator of the food and nutrients available to stream life. It can also provide good cover for macro invertebrate and fish fauna.


The presence of riparian vegetation has a strong influence on the amount of organic matter in a stream as this is the main source of that input. Organic matter is also determined by seasonal leaf litter inputs and the flow characteristics of the stream in terms of how well it retains the organic material. Streams may become over-retentive if they are clogged with aquatic plants or rubbish. Conversely, they may become under-retentive if the stream has very high flows, has been straightened and has no boulders, woody debris etc. for material to be held back by.

Measure it: Select a representative riffle, run and pool and at each of these mesohabitats:

- Visually estimate the percentage of the wetted bed with wood and leaf packs, including trees, branches and roots.
- Calculate the average proportion of the stream-bed where organic matter is present.
- Note down any features of the stream that may affect retentiveness.

	% of the wetted bed with wood and leaf packs	Average proportion of the stream-bed where organic matter is present	Features of the stream that may affect retentiveness
Riffle			
Run			
Pool			

Riparian Vegetation

Vegetation type	Tick box that best suits your stream	Intactness of riparian vegetation
Mature indigenous vegetation with diverse canopy and understorey		<div style="text-align: center;"> <p>High</p>  <p>Low</p> </div>
Regenerating indigenous vegetation in late stage of succession		
Natural, diverse wetland vegetation on banks		
Mature native trees, but damaged understorey		
Mature exotic trees (e.g. Willows and plantation forest)		
Low diversity regenerating bush with stock excluded OR tall exotic shrubs (> 2m)		
Mature flax, long grasses and sedges		
Low diversity regenerating bush with stock access OR Early stage restoration planting OR Short exotic shrubs (< 2m) OR Immature plantation forest		
Mainly long grasses (not grazed or mown)		
Grazed wetlands		
Mainly short grasses (grazed or mown)		
Disturbed bare soil or artificial surfaces		

NB: Use photographic examples from the SEV Pages 41-43...as shown over page



Mature indigenous vegetation



Regenerating indigenous vegetation



Natural wetland vegetation



Native trees with damaged understory



Mature exotic trees



Low diversity regenerating bush



Mature flax



Early stage restoration planting



Long grasses



Grazed wetland vegetation



Short grasses (mown)



Artificial surface

Stage Two

Water Quality – non-biological tests



A bit of background: Water clarity is an indirect measurement of the amount of suspended solids in water. As erosion occurs within a catchment, tiny particles of clays, silts or small organic particles are washed into waterways. These particles can be held in the water current and are called suspended solids. The faster the water is moving the more suspended solids it can carry. If soils enter the stream in unnatural quantities the whole system is quickly thrown out of balance.

Soil type in the catchment can affect water clarity. Suspended material restricts light passing through the water column. Reduced light limits natural and healthy plant growth, which in turn affects the aquatic life relying on those plants for food.

Test it: Collect a litre of water from an undisturbed area of stream and pour it into the clarity tube. Place the magnet in the tube and put the lid on. Move the black disk away from you until it disappears (when the edge of the black disk is no longer decipherable). Note this distance. Then slowly move the disk along the tube back towards you until it reappears. Note this distance too. On the outside of the tube: read the distance measured from the front of the disk to the clear viewing end. Repeat to obtain at least two further readings. Get another person to take a reading if possible. Before each reading, gently agitate the water column to ensure that any sediment stays in suspension. (Don't shake so hard that lots of bubbles are produced in the water, because these will reduce the sighting distance.) Take the average of all the readings to obtain the average clarity.

Clarity =



Conductivity =

A bit of background: Conductivity is the ability to conduct electricity. Water conducts electricity because it contains dissolved solids that carry electrical charges. Therefore, measuring the conductivity of the water indirectly measures the amount of substances such as calcium, bicarbonate, nitrogen, phosphorus, iron, or sulphur dissolved in water, i.e. the amount of Total Dissolved Solids (TDS) in the water. This is important as a water quality measure because most streams have a fairly constant range of conductivity under normal circumstances. Consequently, if a significant change in conductivity is detected then this can be an indicator that a discharge or some other source of pollution has entered the water.

Test it: A conductivity meter is used to measure conductivity. It applies voltage between two electrodes. The drop in voltage between the two electrodes is used to measure the resistance in the water. This is converted into conductivity. The conductivity meter is placed in the water and allowed to stabilise before a reading is taken. Conductivity is measured with a meter in milli siemens per metre units (mSm^{-1}). The natural conductivity of fresh water varies from very low values (less than 3.0 mSm^{-1}) to very high values 25 or more mSm^{-1}), which may be unsuitable for irrigation. The average for New Zealand rivers is 8.5 mSm^{-1} .



Temperature =

A bit of background: *Measuring water temperature is very simple and is a very important factor in water quality.*

Most creatures can survive in water temperatures ranging between 10 degrees Celsius and 25 degrees Celsius. If the temperature rises or falls outside that range, the creatures may die or move away.

The influence of temperature on water quality is significant. Many of the physical, chemical and biological characteristics of streams are directly affected by temperature.

Temperature influences:

- o the amount of oxygen that can be dissolved in the water
- o the rate of growth by algae and larger water plants.
- o the sensitivity of organisms to toxic wastes, parasites and diseases.
- o the structure of aquatic communities.

Several things determine water temperature in a stream but the most important are weather, season, shading, water depth and inflows from other sources.



DTH – Camellia Neilson

Test it: Spot temperature readings are simple to make with thermometers. Alcohol (rather than mercury) thermometers are most suitable for field use. Because stream water temperature varies significantly throughout the day, a spot temperature reading of the maximum temperature on a clear sunny day in the mid-to-late afternoon is best. To avoid localised high-temperature bias,

Flow (Velocity)

Measure the water depth twice at the fastest point on each cross section using a flat bladed metal ruler. For the first measurement, position the ruler parallel to the current and record the depth (d_1). For the second measurement, turn the blade so that it is perpendicular to the current and a 'bow wave' forms on the upstream face of the ruler. Record the depth at the top of this bow wave (d_2). The difference in these two depth measurements ($d_2 - d_1$) can be used to calculate the water velocity within 10% of flow meter readings.

If the difference ($d_2 - d_1$) is less than 2mm, then the usefulness of the method is compromised. In such cases, measure the distance a floating particle travels in a fixed time period (commonly 10 seconds). Therefore, for each of your cross sections, you will have either a measurement based on either the ruler method or the floating particle method.

These measurements are used to estimate the maximum flow velocity (m/s) at each cross section;

The velocity estimate (v) based on the ruler method is calculated as, $v = \sqrt{196 \times (d_2 - d_1)}$.

The velocity estimate (v) based on the floating particle method is calculated as, $v = \text{distance travelled} / \text{time taken}$

- V_{veloc}
 - the mean of the ten velocity estimates is used to calculate V_{veloc}

V_{veloc} Scoring Table

Cross Section	Ruler Method	Floating Particle Method:		Velocity
	(d ₂ -d ₁) mm	Distance Travelled (m)	Time taken (s)	estimate
1				
2				
3				
4				
5				
6				
7				
8				
9				
10				
			Mean Velocity	

Dissolved Oxygen

A bit of background: Dissolved oxygen (DO) refers to the small amount of oxygen gas dissolved in the water. Dissolved oxygen is vital for life in freshwater environments. Low levels of DO directly affect fish, macroinvertebrates, micro-organisms and plants all of whom depend on it for the process of respiration, i.e. to breathe. High levels of DO may indicate anoxic (too much respiration) or eutrophic (too much productivity) conditions. This can be similarly harmful for stream life and its healthy functioning.

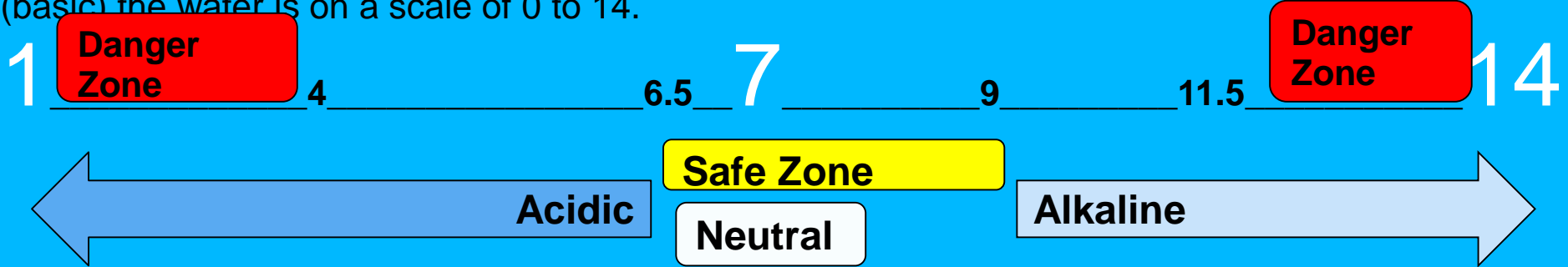
Test it: Place a dissolved oxygen (DO) probe in gently flowing water – ideally at the downstream end of a pool. Ensure the DO meter is calibrated before use according to manufacturer's instructions. Allow the value to stabilise and record DO % saturation and water temperature. A reading as close to dawn as possible provides a measure of the minimum DO saturation in-stream, and a reading in the late afternoon provides a measure of the maximum DO concentration in-stream.

If you don't have access to a DO meter there are some simple visual observations that you can make that can assess likely DO levels in your stream. This assessment has been taken and simplified from the Stream Ecological Valuation (SEV) methodology (Storey *et al.* 2011). Tick the box which best describes the indicators of oxygen reducing processes that are present in your stream reach.

Status		Tick box that best suits your stream	DO levels
Optimal	No anaerobic sediment No odours or bubbling when sediment disturbed Little or no macrophyte biomass (summer), or no areas of slow flow, low shade and soft substrate (winter)		High
Sub-optimal	No anaerobic sediment Some bubbling when sediments are disturbed, but no odour Moderate macrophyte biomass (summer), or moderate areas of slow flow, low shade and soft substrate (winter)		
Marginal	Small patches of anaerobic sediment Some bubbling and sulphide odour when sediments are disturbed Some sewage fungus may be present Dense macrophyte biomass (summer), or no areas of slow flow, low shade and soft substrate (winter)		
Poor	Much black anaerobic sediment Extensive bubbling with sulphide odour when sediments disturbed Surface scums present Abundant sewage fungus may be present		Low



A bit of background: The pH test for water is a measure of how acidic or alkaline (basic) the water is on a scale of 0 to 14.



Pure water has a neutral pH and is 7 on the pH scale. pH measurements below 7 indicate the water is acidic. pH from 7 to 14 indicates the water is alkaline.

Animals and plants in streams are adapted to certain ranges of pH. Creatures prefer pH levels between 6.5 and 9 and changes in pH outside the normal range of a water body will cause more sensitive species to die as it changes other aspects of the water chemistry or the creature's biochemistry.

A pH of below 4 or above 11.5 is sufficient to kill all fish in the water. There is a gradation of effect up to these extremes remembering that for every one unit change on the pH scale, there is approximately a ten-fold change in how acidic or alkaline the sample is.

pH =

Test it: using the universal indicator paper. Collect a water sample in a small clean jar and then dip the coloured end of the paper into the water for 3 seconds. Then wait 1-3 minutes to approximate the pH according to colour.



Nutrients – phosphate and nitrogen

A bit of background: *A nutrient is a chemical that an organism needs to live and grow and which must be taken in from its environment. Nutrients in streams are used for growth by aquatic plants, periphyton, and microbes (bacteria, fungi).*

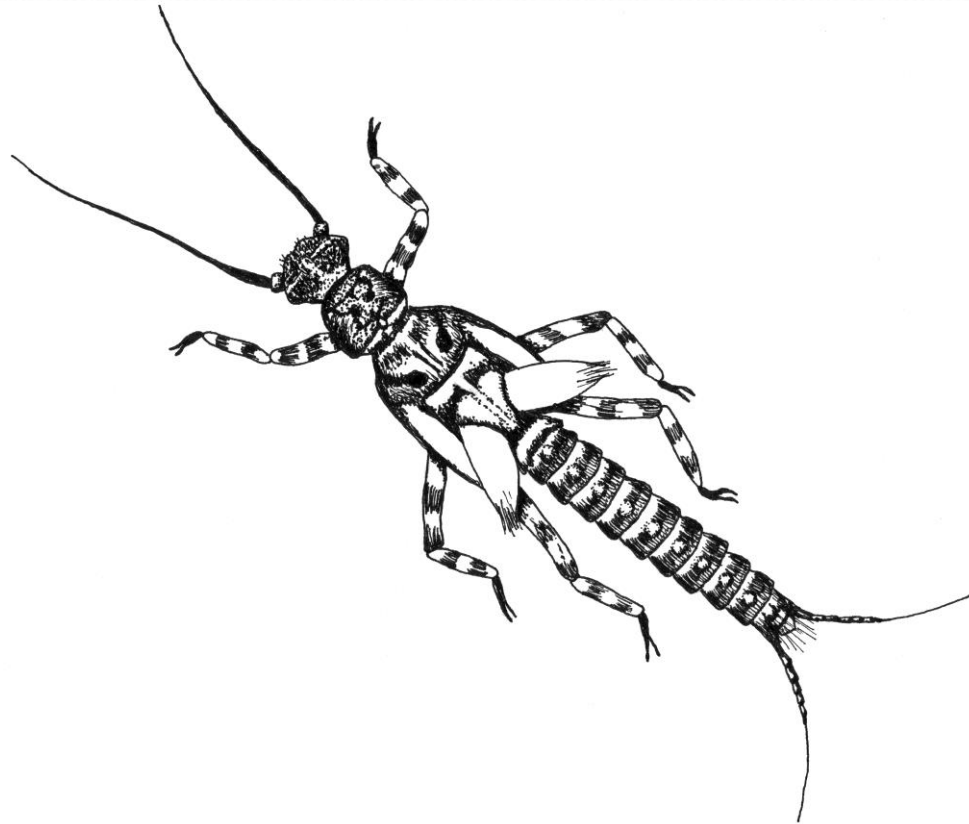
The two primary nutrients in water are nitrogen (N) and phosphorus (P). Human activities can result in external inputs of nutrients entering streams (e.g. fertiliser leachate/runoff, industrial wastewater, urban stormwater and sewage discharges). These are often at high concentrations and can stimulate nuisance growths of aquatic plants, periphyton, and microbes. These inputs can be from point sources (e.g., pipes, drains) or diffuse sources (e.g., groundwater seepage, runoff).

When nitrogen or phosphorus levels exceed normal levels in a stream, the balance in the ecosystem is lost and excessive plant growth takes place. This is a process called eutrophication and can cause problems such as algal blooms and growths. This is further exacerbated when these plants die, as oxygen is used in their decay. Lower oxygen levels reduce the diversity of macroinvertebrates that the stream can support and in extreme cases can kill fish.

Test it: Water samples can be collected for nutrient analysis and assessed in a lab. The water sample should be collected in containers provided by the lab, and by using a grab to take the sample from the centre of the stream channel during a period of stable river flow. Store the water sample chilled during transport to the laboratory. Samples should be processed within 24 hours of collection or frozen to be analysed later.

Stage Three

Testing the life – in stream biota



Bacteria

A bit of background: Some water quality tests measure micro-organisms in the water to ensure water is safe for swimming or shellfish collecting. Faecal microbes that can cause disease in humans are some such micro-organisms and high levels of faecal material can indicate the possible presence of these bugs. Faecal contamination is normally monitored using “indicator” microbes that are universally present in the faeces of warm-blooded animals (including people). These are the bacterium *Escherichia coli* (in freshwaters) and the Enterococci group (in marine waters).

Test it: Water samples can be collected for *E.Coli* bacteria analysis and assessed in a lab. The water sample should be collected in containers provided by the lab, and by using a grab to take the sample from the centre of the stream channel during a period of stable river flow. Store the water sample chilled and out of sunlight (UV light kills bacteria) during transport to the laboratory. Samples should be processed within 24 hours of collection or frozen to be analysed later.

Periphyton

A bit of Background:

Periphyton are sessile (stationary) freshwater organisms that are attached to, and grow on, surfaces on the stream bed, i.e. rocks, sand, wood and macrophytes (instream plants). They mainly comprise algae, but can also be bacteria and fungi. Periphyton can take two general forms:

1. microscopic, single-cell algae forming thin layers on stream substrates (i.e., diatoms). These are more common in fast-flowing, higher gradient streams with lower light levels.
2. macroalgae that develop as filaments, sheets or mats. These are more common in low-gradient, open streams with higher light levels.

Periphyton is an important part of the food chain. It is a primary producer that photosynthesizes and is one of the main food sources for stream fauna such as macroinvertebrates (e.g. snails, mayflies, caddisflies, midges), which in turn are fed upon by fish and birds.

Since algae require light to photosynthesize and grow, light levels are the primary factor controlling periphyton abundance. It is also regulated to a lesser extent by stream flow and scour regime (which can dislodge it), nutrients, temperature, and grazing.

In high light conditions, sluggish, low-gradient streams with no riparian cover and run-off from a pastoral catchment, nuisance growths of periphyton are likely to develop as mats or sheets across the stream bed and make the habitat unsuitable for many sensitive invertebrate species. Conversely, in higher-gradient, fast flowing, shaded streams in a forested catchment, a thin covering of microscopic algae may be present on the rocky substrate providing a good food resource for resident macroinvertebrates.

Test it:

The form is provided overpage. This method assesses periphyton cover across 5 evenly spaced transects at the reach scale (50–100 m). These could be the same as the substrate measurement transects.

Begin at the downstream transect. For each transect, assess periphyton cover in a 10 cm diameter circle at 5 evenly spaced sampling points across the transect (i.e., at 10, 30, 50, 70 and 90% of the width).

Assess periphyton on whatever substrate occurs at each point (i.e., cobbles, sand, wood, macrophytes). Record the percentage cover of upper surface for the different periphyton categories (which are based on mat thickness or filament length and colour;

Repeat for all five transects. Calculate a mean % cover for each periphyton category across the 5 transects.

Assess periphyton during the growing season (summer and autumn when plant biomass peaks) under base flow conditions and at least 3 weeks after a high flow event.

Data sheet for periphyton rapid assessment.

Stream name:

Date:















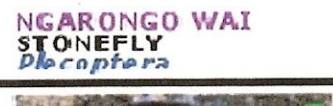



Transect #:

Thickness category	Colour category	Transect 1	Transect 2	Transect 3	Transect 4	Transect 5	Mean % cover
Thin mat/film (<0.5mmthick)	N/A						
Medium mat (0.5-3mm thick)	Green						
	Light brown						
	Black/dark brown						
Thick mat (>3mm thick)	Green/light brown						
	Black/dark brown						
Short filaments (≤ 2 mm long)	Green						
	Brown/reddish						
Long filaments (>2cm long)	Green						
	Brown/reddish						
Submerged bryophytes	NA						
Iron bacteria growths	NA						

Briefing for macroinvertebrate sampling:

- Choosing your site
- Use of and preparing the equipment
- Sampling the different microhabitats - Kick Sampling, Scoop Netting, Picking up rocks
- Identification and sensitivity scores
- Reinforce Kaupapa – how to take care of your findings
- Reinforce how to keep safe – whistle, take care, only go in up to knees, keep together.
- Ensure everyone is in small groups with required adult:child ratios
- Outline the time they have and when to regroup – what is expected then e.g. clean out gear and bring worksheets back for sharing.

CREATURE ID CHART

 <p>5</p> <p>6mm</p> <p>TE PEPEKE KAUKIORE BACKSWIMMER <i>Nataneptidae</i></p>	 <p>5</p> <p>25mm</p> <p>KIHITARA DAMSELFLY <i>Xanthocnemis</i></p>	 <p>5</p> <p>10-30mm</p> <p>NGARONGO WAI STONEFLY <i>Plecoptera</i></p>	 <p>9</p> <p>20mm</p> <p>PIRIWAI COMMON MAYFLY <i>Deleatidium</i></p>	 <p>7</p> <p>30mm</p> <p>NGARO PARIRAU DOBSONFLY <i>Archibaulia</i> etc</p>	 <p>5</p> <p>6mm</p> <p>TE KAIHOE POTI WATER BOATMEN <i>Corixidae</i></p>
 <p>6</p> <p>30mm</p> <p>KAPOAT DRAGONFLY LARVAE <i>Petaluridae urupetala</i></p>	 <p>3</p> <p>10mm</p> <p>NGATA FRESHWATER SNAIL <i>Potamopyrgus</i></p>	 <p>6</p> <p>120mm</p> <p>KAKAHI FRESHWATER MUSSEL <i>Hyridella menziesi</i></p>	 <p>5</p> <p>150mm</p> <p>KOURA FRESHWATER CRAYFISH <i>Paranephrops planifrons</i></p>	 <p>5</p> <p>30mm</p> <p>PĀPAKA FRESHWATER CRAB <i>Halticarcinus / Helice</i></p>	 <p>5</p> <p>30mm</p> <p>KOURAURA FRESHWATER SHRIMP <i>Paratya curvirostris</i></p>
 <p>6</p> <p>15mm</p> <p>NGARO WAIWAI ANGA STONY-CASED CADDIS <i>Pynocentria</i></p>	 <p>5</p> <p>20mm</p> <p>NGARO WAIWAI ANGA WOODY-CASED CADDIS <i>Triplectides</i></p>	 <p>5</p> <p>15mm</p> <p>NGARO WAIWAI KORE ANGA FREE LIVING CADDISFLY <i>Hydrobiosis</i></p>	 <p>3</p> <p>7mm</p> <p>LEECH <i>Glossiphonia</i></p>	 <p>1</p> <p>30mm</p> <p>TOKE ULIGOCHAETE WORM <i>Eisenella</i></p>	 <p>2</p> <p>20mm</p> <p>MIDGE LARVAE <i>Chironomidae</i></p>

SENSITIVITY ■ High ■ Moderate ■ Low

SENSITIVITY SCORE **10** = Highest Sensitivity
1 = Lowest Sensitivity

Chart created for WBC by Ting Yiu 2011

Photographs by Kim Jones

Illustrations by Stephanie. J. Bowman www.bowmansart.com

Debrief after waterway study:

- Do a head count
- Wash or sanitise hands
- Ensure you have all stream equipment
- Make sure students have checked, cleaned & dried all stream equipment – prevent the spread of any unwanted organisms.
- Ensure students have returned all life to the waterway.
- Go over what was found - using opportunity to record all in stream biota (over page)
- How healthy do people think the stream is now? Has their perspective changed? If so, why?
- What actions could be done to improve the health of the stream?
- Which of those actions could this group contribute to?
- Go over the objectives of the day – did we answer our questions or do we need to do more research? Have we now got even more questions??
- Talk about what is next – follow up visit – who needs to prepare what.

Questions

- Give students and adult's time to ask questions

After students leave site:

- Ensure all gear has been picked up from the study site.
- Leave the area as or better than you found it e.g. pick up rubbish etc.
- Check, Clean and Dry all of the equipment before next use
- Create a photo story with the pictures and results of the day and set a date to use it for the next follow up visit.