River Monitoring Guidelines

Environment Agency
Shropshire Wildlife Trust
a co-operative council
Telford & Wrekin Council
Severn Trent Water

Contents

Acknowledgements

Information included in this handbook comes from a variety of sources and has been referenced where appropriate. Please do not reproduce information from this handbook for purposes other than undertaking river monitoring. Many thanks go to the Love Your River Telford volunteers and funders for their support and dedication during the project.

Overview

This handbook is designed to provide and understanding of the practical skills the Love Your River team gathers during their on-ground training days and the reasoning behind the suggested methods. It forms part of an essential resource pack that equips volunteers with skills and resources to undertake monitoring on rivers, streams and other waterbodies.

The pack includes recording sheets and other useful information, and links to supplementary resources are available through the Shropshire Wildlife Trust website (www.shropshirewildlifetrust.org.uk/what-we-do/wild-water).

Monitoring of this kind provides a snap-shot in time and can give an indication of the current health of the waterway. To build a better picture a longer time frame of monitoring is needed. Returning to the same section of waterway over several years will give an understanding of whether the waterway is improving, declining or staying the same over time.

River monitoring should only be carried out if it is safe to do so. If the water is high or fast flowing it may be unsafe to collect samples or conduct a bankside survey. Equipment such as collection cups on long poles may reduce these risks.

Freshwater invertebrate monitoring

Overview

The presence/absence and abundance of certain freshwater invertebrates (often called macroinvertebrates) – the aquatic larval stage of various insects - can tell a story of a water body's health. Because some invertebrates are more sensitive to pollutants than others, we can use them as "indicator species". Assessing the types of invertebrates found indicates something about the health of the water body they have been found in. For example worms can tolerate high levels of pollutants but some mayflies don't tolerate even low levels of pollution. They also help describe when pollution events occur because some populations take a while to recover if they have been harmed by pollution.

Figure 1 - Volunteers kick sampling on the Reabrook in Shrewsbury

A good strategy for monitoring invertebrates in waterways is to carry out sampling twice a year, once in spring and once in autumn. This will give an indication of seasonal changes. It can also be useful to carry out sampling before and after any major changes in the catchment such as construction of buildings or habitat improvement projects.

To monitor the invertebrates present in our waterways, we assign a score to them. There are various standardised methods for doing this type of scoring. Some require advanced knowledge of invertebrate identification, the use of microscopes, and the dissection of different parts of the animal. Our aim is to be able to collect samples and identify the invertebrates present as accurately as possible in the field. In light of that, we have compiled a list of common aquatic invertebrates and assigned a score to each one that reflects how tolerant it is to pollution based on the Whalley Hawkes Paisley Trigg (WHPT) Method for assessing river invertebrate communities. Once we have identified and counted each invertebrate present in the sample and added up the scores we get an indication of how healthy that waterway is.

Kick Sampling

The typical sampling method for streams and rivers is called kick sampling and involves disturbing the material on the bed of the river or stream and collecting the freed organisms in a net. Because invertebrates use a variety of habitat types it is important to collect samples from the different habitats present such as fast moving riffles, shallow water, slow water, weeds and tree roots. This increases the chances of collecting all the different species present at the site.

The kick sample should be conducted for three minutes. Where possible, the time should be divided proportionally between the different habitats depending on their abundance. For example, if riffles occupy half (50%) of the site they will be sampled for half of the time (= 90 seconds), and if vegetation occupies a quarter (25%) of the site it will be sampled for a quarter of the time (= 45 seconds).

Figure 2 - LYR volunteer kick sampling on the Nedge Brook in Telford

Place the net on the riverbed and disturb the area just upstream of the net by kicking or shuffling through the substrate, for the time allocated. The animals will then be carried downstream by the current into the net. Avoid kicking coarse debris into the net. Any debris caught in the net should be removed, while making sure to rinse the invertebrates that are clinging to it back into the net.

For in-stream vegetation and tree roots, sweep the net through the area for the allocated time, trying to scrape the net against the debris. It is also advisable to carry out an additional one minute hand search of large stones by gently rubbing the stones in the water, letting any animals be carried downstream into the net. Be careful as there may be glass, metal or other sharp objects on the riverbed.

In order to identify what has been collected during the kick sample, fill a tray with river water to a depth of a couple of centimeters and lower the net into the water. Carefully turn the net inside out, and shake gently to release the contents. If you have collected a large sample or lots of debris, it may be necessary to examine the contents by taking smaller portions at a time. To do this you will need to empty the contents of the net into a bucket half filled with water. Remove a sample from that bucket using a kitchen sieve or similar, and empty the contents into your tray. When you have finished examining the sample, empty the contents into a second bucket or put it back into the river. Continue taking sub-samples until your first bucket is empty.

Figure 3 - Volunteers identifying aquatic invertebrates from kick samples

It may be necessary to remove each invertebrate into a separate tray using a small spoon as this will make identification easier. It may not be possible to count every individual present if there are lots. In this case the abundance should be estimated into the abundance groupings (greater than or less than 10, greater than or less than 100 or greater than or less than 1000). Take care when removing individuals as some bite!

Once all the animals have been identified, return the sample to the river, ideally in the same location as where the sample was collected from.

Identification

In order to ensure that data collected is reliable and useful we use a standardised method for identifying and recording the invertebrates collected in each sample.

The Field Studies Council publication “A key to the major groups of British freshwater invertebrates” provides a clear and easy to follow identification key. All the target species listed in the appendix that have been selected for scoring are present in this guide book.

Chemical analysis

Overview

Certain chemicals are harmful to plants, animals and humans. When these chemicals are present in our waterways they can be detected using a variety of scientific methods. Sometimes there are visual clues that there is something present in the water that is out of the ordinary such as detergent smells or bubbles, strange colours, or the presence of dead fish. Sometimes the water will look clean and healthy but analysis can show that levels of certain chemicals are too high for certain plants and animals to tolerate.

Regular monitoring can help us understand how the chemistry of the water changes both within a season and between different seasons and therefore how to identify unnatural levels of chemicals.

Figure 4 - collecting water samples for further analysis on the War Brook

Sampling Methods

A standard water monitoring kit will contain:

- test strips for ammonia, nitrate and pH
- a temperature probe (or a combined temperature, conductivity, pH probe)
- sample bottles
- sampling pole with attached sample cup
- gloves
- pencil and note pad

Results should be recorded either on a printed record sheet or in the note pad provided and transferred onto the Excel spreadsheet once complete (available from the Trust website).

Ammonia and Nitrate

Nitrates and ammonia are essential plant nutrients but elevated levels can cause significant water quality problems. They can cause excessive plant growth which uses up the dissolved oxygen available, making the water toxic for other aquatic live.

Sources of nitrates include sewage, runoff from fertilised lawns and parkland, run-off from agricultural land, and industrial discharges. Ammonia can also find its way into watercourses from misconnected properties where, for example, plumbing for washing machines is incorrectly connected to surface water drains that discharge into nearby rivers, streams and canals.

How to measure ammonia and nitrate

Collect a small sample of water from the waterbody you are monitoring. Use the test strips, ensuring you return the lid to the test strip bottle. Dip the strip into the water sample, moving it up and down within the sample for 30 seconds.
Allow the test strip to sit for an additional 30 seconds before using the colour chart on the bottle to determine the level of ammonia or nitrate present in the water sample. The amount of ammonia and nitrate is measured in parts per million (ppm). Do this using the individual strips for ammonia and nitrate and record these values on the record sheet.

Figure 5 - Matching up the colour of the test strip with the colour coding on the bottle

pH

pH is a measure of the alkalinity or acidity of a substance. This is measured on a scale from 1 to 14. Lower pH values indicate more acidic conditions while higher pH values indicate more alkaline (or basic) conditions. pH affects many chemical and biological processes in the water. Low pH can allow toxic elements in the water to become mobile. This can produce conditions that are toxic to aquatic life, particularly to sensitive species. Most aquatic animals prefer a range of 6.5 - 8.0. pH outside of this range reduces the biodiversity in the watercourse.

How to measure pH

Collect a small sample of water from the waterbody you are monitoring. Use the paper test strips by dipping the strip into the water sample and removing it immediately. Allow the test strip to sit for a few seconds before using the colour chart on the container to determine the pH of the water. Record this value in the record sheet. If using the pH probe, insert the probe into the water sample or directly into the water body. Keep the probe tips fully submerged in the water until the read-out on the probe is stable. Record this value in the record sheet.

Temperature

Changes in temperature impact plants and animals that are adapted to living within specific temperature ranges and can make them more sensitive to parasites and disease. As temperature increases, oxygen levels decrease which can also adversely affect organisms.

The temperature of a watercourse will vary naturally depending on many factors including the width and depth of the water body. Shading from vegetation will significantly alter the temperature of the water, particularly in the summer months. Causes of temperature change include weather, removal of vegetation and other shade providers, impoundments (a body of water confined by a barrier, such as a dam), discharge of cooling water, urban rain/storm water, and groundwater inflows to the stream.

How to measure temperature

Temperature can be recorded directly from the water body you are monitoring. If there is no safe access to the water, collect a small sample of water using a long pole with a cup on the end or a similar piece of equipment. Insert the temperature probe into the water sample or directly into the water body. Keep the probe tips fully submerged in the water until the read-out on the probe is stable. Temperature is measured in degrees Celsius (°C) or degrees Fahrenheit (°F). Record this value in the record sheet.

Figure 6 - Combined temperature, pH and conductivity probe

Conductivity

Conductivity is a measure of the ability of water to pass an electrical current. Conductivity in water is affected by the presence of inorganic dissolved solids such as chloride, nitrate, phosphate and sodium. Organic compounds like oil and alcohol do not conduct electrical current very well and therefore have a low conductivity when in water.

Conductivity in streams and rivers is affected primarily by the geology of the area through which the water flows. Streams that run through areas with granite bedrock tend to have lower conductivity because granite is composed of more inert materials. Streams that run through areas with clay soils tend to have higher conductivity because of the presence of conductive minerals that are washed into the water from the clay.

Discharges of pollution into streams can change the conductivity depending on their make-up. Sewage from a missed plumbing connection would raise the conductivity because of the presence of chloride, phosphate, and nitrate; an oil spill would lower the conductivity.

Conductivity is measured in microsiemens per centimeter (µs/cm). The conductivity of rivers generally ranges from 50 to 1500 µs/cm. Rivers supporting good aquatic life have a range between 150 and 500 µs/cm. Conductivity outside this range could indicate that the water is not suitable for certain species.

How to measure Conductivity

Conductivity can be recorded directly from the water body you are monitoring. If there is no safe access to the water, collect a small sample of water using a long pole with a cup on the end or a similar piece of equipment.

Using the conductivity probe, insert the probe into the water sample or directly into the water body. Keep the probe submerged in the water until the read-out on the probe is stable. Record this value in the record sheet.

Turbidity

Turbidity is a measure of how cloudy a water sample is. It is the amount of material in the water that limits light passing through. Materials include soil (clay, silt, and sand), algae, plankton and other substances. It is often measured in Nephelometric Turbidity Units (NTU). Some water bodies are naturally turbid, or turbid at certain times, while others are not.

Higher turbidity increases water temperatures because the materials in the water absorb more heat. This in turn reduces the concentration of dissolved oxygen (DO) because warm water holds less DO than cold. Suspended materials can clog fish gills, reducing resistance to disease in fish, lowering growth rates, and affecting egg and larval development. As the particles settle, they can blanket the stream bottom, especially in slower waters, and smother fish eggs and invertebrates.

Turbidity can be useful as an indicator of the effects of runoff from construction, urban environments, agricultural practices, discharges, and other sources.

Regular monitoring of turbidity can help detect trends that might indicate changes such as increasing erosion in developing areas. However, turbidity is closely related to stream flow and velocity and should be linked to these factors. Comparisons of the change in turbidity over time, therefore, should be made at the same place and in similar flow conditions.

How to measure Turbidity

There are several technical and expensive pieces of equipment that can be used to measure turbidity. These include spectrophotometers, Secchi tubes and turbidimeters. However a more simple and cost effective method can be used in the field. For this we can use the indicative scale provided at the back of this handbook.

Take a sample of water from free flowing water using a sampling pole and cup. Take care not to stir up debris from the river bed or surroundings. Pour the water into the clear sample pot to about two-thirds full. Look through the water to determine how turbid the water is using the graded scale supplied (measured in NTU). Record the NTU value for how cloudy the water looks. Record whether it is more, less or the same cloudiness compared to the last sample.

Stream Flow Rates

Stream flow rate is the volume of water that moves over a designated point over a fixed period of time. It is often expressed as cubic metres per second (m^3/sec).

The flow of a watercourse is affected by weather, increasing when it rains and decreasing during dry periods. It also changes during different seasons of the year, typically decreasing during the summer months when evaporation rates are high and riverbank vegetation is actively growing and removing water from the ground.

Flow is important because of its impact on water quality and on the living organisms and habitats in the water body. The stream flow rate determines the kinds of organisms that can live in the watercourse (some need fast-flowing areas while others need quiet pools). It also affects the amount of silt and sediment carried by the stream. Sediment in quiet, slow flowing watercourses will settle quickly to the stream bed. Fast moving watercourses will keep sediment suspended for longer in the water. Fast moving watercourses also generally have higher levels of dissolved oxygen because they are better aerated.

How to measure Stream Flow

The speed of the water flowing at the surface of the stream is a simple measurement that gives an approximate indication of the stream flow rate. Additional measurements of the shape of the waterway are needed to calculate the volume of water in the stream.

To calculate the speed of the stream flow, pace or measure out a section of water course. This can be anything from 5 -10 metres depending on the size and flow rate of the water. For faster flowing waterways, longer sections are better. Record the location and length. It is useful to use something permanent like a tree to mark the start and finish points of the section so repeat measurements can be carried out in the future.

Place or drop a readily identifiable, small (approx 5-10 cm) floating object, such as a piece of stick or an orange, into the water at the start of the measured length. Measure the time it takes to reach the end of the section of water. Record these results in a log book. Repeat this 3 or 4 times to get an average.

Understanding the Results

If your water quality sampling shows the following results, please repeat the test:

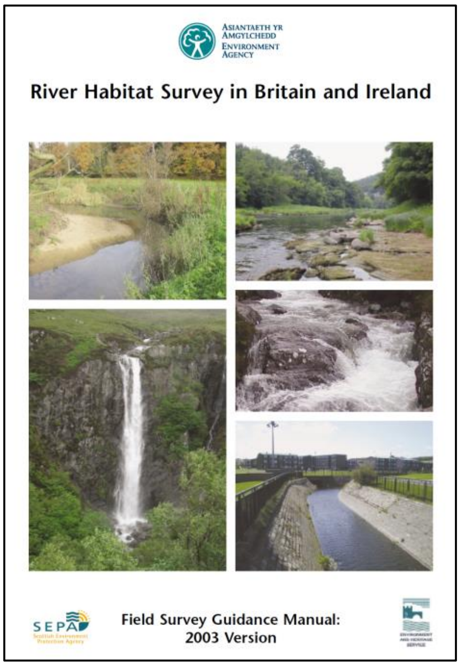
- pH below 6 or above 8
- Ammonia reading above 3.0 ppm
- A change in turbidity that is not thought to be related to weather e.g. following rainfall, warm temperatures
- A temperature higher than 20°C

If, after a second test the results are still the same, please report this to the Environment Agency Clean Stream Team on **0800 807060**. Please also report any pollution incidents to the same number.

River Habitat Survey

Overview

We can learn a lot about our waterways by walking along beside them, looking at how they flow and the land use near them. This information can be used to assess the physical structure of our waterways and help determine the conservation value of the waterway.



Using the River Habitat Survey developed by the Environment Agency means we can record our observations in a systematic, standardised way. The 2003 Field Survey Guidance Manual is a great resource for volunteers as specialist knowledge on geology or botany is not required in order to understand the methodology.

Appendices

The following documents include:

1. Recording sheet for chemical parameters
2. Recording sheet for aquatic invertebrates
3. A list of priority taxa and their abundance scores
4. A guide for estimating turbidity

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Watercourse	Date	Recorder Initials	Grid ref	Time	Temp ^0C	DO	Conductivity	pH	Turbidity	Nitrate	Ammonia	Flow	Weather	Comments

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Taxa	Total present in sample	Abundance category (AB1 - AB4)	Score

Number of Taxa	
Score	
Average Score Per Taxa Sum of scores for all taxa ÷ number of different taxa	

AB1 = 1-10
AB2 = 11-100
AB3 = 101-1000
AB4 = >1000

	Common name	Scientific name	AB1	AB2	AB3	AB4
Crustacea						
	Freshwater shrimp	*Gammaridae*	4.2	4.5	4.6	3.9
	Freshwater hog louse	*Asellidae*	4.0	2.3	0.8	-1.6
	Freshwater crayfish	*Astacidae*	7.9	7.9	7.9	7.9
Bivalves						
	Swan or duck mussel	*Unionidae*	5.2	6.8	6.8	6.8
	Orb cockle or pea mussel	*Sphaeriidae*	4.4	3.5	3.4	2.3
	Zebra mussel	*Dreissenidae*	3.7	3.7	3.7	3.7
Snails (Gastropods)						
	Freshwater limpet	*Ancylidae*	5.8	5.5	5.5	5.5
	Pond snail	*Lymnaeidae*	3.6	2.5	1.2	1.2
	Ramshorn snail	*Planorbiidae*	3.2	3.0	2.4	2.4
True Flies						
	Blackfly	*Simuliidae*	5.5	6.1	5.8	3.9
	Meniscus midge	*Dixidae*	7.0	7.0	7.0	7.0
	Non-biting midge (Bloodworms)	*Chironomidae*	1.2	1.3	-0.9	-0.9
	Biting midge	*Ceratopogonidae*	5.4	5.5	5.5	5.5
	Mosquito	*Culicidae*	2.0	1.9	1.9	1.9
	Rat-tailed maggot	*Syrphidae*	1.9	1.9	1.9	1.9
	Crane fly larvae and their relatives	*Tipulidae*	5.4	6.9	6.9	7.1
Leeches						
		Piscicolidae	5.2	4.9	4.9	4.9
		Glossiphoniidae	3.4	2.5	0.8	0.8
		Hirudinidae	-0.8	-0.8	-0.8	-0.8
Water Bugs						
	Water measurer	*Hydrometridae*	4.3	4.3	4.3	4.3
	Pond skater	*Gerridae*	5.2	5.5	5.5	5.5
	Water scorpion	*Nepidae*	2.9	2.9	2.9	2.9
	Greater water boatman	*Notonectidae*	3.4	3.9	3.9	3.9
	Lesser water boatman	*Corixidae*	3.7	3.9	3.7	3.7
Water Beetles						
	Whirligig beetle	*Gyrinidae*	8.1	9.0	9.0	9.0
	Diving beetle	*Dytiscidae*	4.5	4.8	4.8	4.8
	Water scavenger beetle	*Hydrophilidae*	5.8	8.8	8.8	8.8
Mayflies						
	Swimming mayfly	*Baetidae*	3.6	5.9	7.2	7.5
	Burrowing mayfly	*Ephemeridae*	8.3	8.8	9.4	9.4
	Flattened or flat-headed mayfly	*Ecdyonuridae*	8.5	10.3	11.1	11.1
Dragonflies						
	Dragonfly	*Anisoptera*	6.2	6.2	6.2	6.2
Damselflies						
	Damselfly	*Zygoptera*	5.1	5.3	5.3	5.3
Caddisflies						
	Finger net caddisfly	*Philopotamidae*	11.2	11.1	11.1	11.1
	Trumpet net/tube net caddisfly	*Polycentropodidae*	8.2	8.1	8.1	8.1
	Net spinning caddisfly	*Hydropsychidae*	5.8	7.2	7.4	7.4
	Purse-case caddisfly	*Hydroptilidae*	6.1	6.5	6.8	6.8
	Cased caddisfly	*Leptoceridae*	6.7	6.9	7.1	7.1
		Phryganeidae	5.5	5.5	5.5	5.5
Stoneflies						
	Stonefly	*Plecoptera*	10.5	11.3	11.4	11.4
Worms						
	True worm	*Oligochaeta*	3.6	2.3	1.4	-0.6
Flatworms						
	Flatworm	*Triclada*	3.5	3.7	3.7	3.7

Turbidity (NTU)