Biological assessment

Version: June 2018

Aquatic macroinvertebrate sampling, processing and index calculation

1 Purpose and scope

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

2 Associated documents

Sampling design and preparation:

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

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

3 Health and safety

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

4 Permits and approvals

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

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

5 Skills, training and experience

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

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



competency.

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

6 Equipment

See Appendix 1 for example equipment checklist.

7 Procedure

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

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

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

7.1 Habitat selection

7.1.1 Sampling site and aquatic habitat

The site is defined as 50m upstream and 50m downstream of the entry point (total of 100m of the stream reach). Aquatic habitats are defined using a combination of visual identification and velocity to depth ratio validation. The key visual identifiers for each habitat type are summarised in Table 1. The velocity to depth table (Appendix 2) can be used to confirm the identification of a habitat type based on flow. Velocity should be calculated using a flow meter at several points along a stream cross section. Alternatively a visual estimator can be used, where the time it takes for an object to move down a known length of stream is measured. Depth can be measured using a depth measuring pole.

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

Table 1: Key descriptors of different habitat types

Habitats to be sampled in each region to align with the EPP (Water) WQOs for macroinvertebrates are summarised in Table 2.

Table 2: Habitats to be sampled in each region to align with the EPP (Water) Water Qua	ality Objectives
(WQOs)	

Areas with aquatic macroinvertebrate water quality objectives (WQOs) ¹	Habitats assessed
Wet Tropics ^a	Edge and riffle
Fitzroy Basin waters	Edge and composite of all bed habitats
Mackay-Whitsundays ^b	Edge and riffle
Capricorn-Curtis Coast region ^c	Edge
Moreton Bay/south-east Queensland	Edge

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

7.2 Sampling aquatic macroinvertebrates using a dip net

7.2.1 Preparation

1. Check sampling area is of a wading depth i.e. maximum depth of 1–1.5m. For Workplace Health and

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

Safety reasons, ensure it doesn't have a strong current and isn't slippery.

- 2. Check nets for damage or holes. Only use nets that are free of damage or holes.
- 3. Wash nets and buckets prior to sampling each site to remove animals left from previous sampling.
- 4. Record site information (for example site location, site code, date, time etc.).
- 5. Record GPS co-ordinate of site.

7.2.2 Sampling habitats

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

7.2.2.1 Edge/backwater

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

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

7.2.2.2 Riffle/runs

- 1. While holding the net with its mouth facing upstream, disturb the substratum with the feet so that macroinvertebrates are washed into the net. Turn and rub cobbles and rocks by hand to dislodge organisms.
- 2. Continue this process walking backwards and working upstream, covering both the fastest and slowest flowing sections of the riffle/runs until a distance of 10m is sampled.
- 3. Place several cobbles into the net and then transfer them into a bucket for direct picking of animals from the cobbles.

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

7.2.2.3 Pools

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

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

7.2.2.4 Composite bed

1. Sample 5m of each available bed habitat at the site, one after the other into the one net, using the

methods described in Section 7.2.2.2 and 7.2.2.3.

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

7.2.3 Completion of sampling

7.2.3.1 For live picking of sample

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

7.2.3.2 For laboratory picking of sample

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

Project·Name:	///////	_/¶
Site Name:		¶
Site No:	·Habitat:	¶
Collector:		¶
Collecting Method:		¶
Picking Method:		A

Figure 1: Label for laboratory picked sample

7.3 Live picking aquatic macroinvertebrates

7.3.1 Sample preparation

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

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

7.3.2 Method

- 1. Set up table and chairs with timer, white trays, picking tool and vial or jar half filled with 70% ethanol or methylated spirits. For one of the first three samples collected in a sampling program, the procedure in Section 7.3.4 must be followed.
- 2. Place a small amount of the sample material and water into a white tray.

- 3. Start the timer.
- 4. Pick aquatic macroinvertebrates from the tray using tweezers, spoon and pipettes, and transfer into the vial with the ethanol (or methylated spirits). Suck up small taxa, e.g. mites, with a pipette.
 - Aim to collect only 10 individuals of any one taxon, i.e. do not spend a large proportion of picking time concentrating on animals that all appear to be the same. When uncertain of the identity, then collect all of the organisms. At least 30 midge larvae (Chironomidae) should be collected to ensure adequate representation of the sub-families. The aim of live-picking is essentially to collect the greatest number of taxa as possible from the sample. Ten individuals of apparent taxa provides some indication of abundance and assists in collecting taxa that are visually similar.
 - At the start of the live pick, the common and abundant taxa should be targeted for the first 5 minutes. After that, the major picking effort should be directed at finding the less common, inconspicuous taxa.
- 5. Work progressively through the sample, replacing picked material with fresh sample.
- 6. After picking for 30 minutes, search specifically for new taxa for 10 minutes. If any new taxa are found in these 10 minutes, extend the picking time by another 10 minutes.
- 7. Follow this procedure until either no new taxa are found or a maximum of 60 minutes have been spent on picking.
- 8. Record the picking time, and add to the vial label (Figure 2).
- 9. Record if the sample has very few animals and any likely reasons why that may be so (e.g. very silty/sandy bed). Add note to vial label (Figure 2).

Note:

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

Live Picked Macroinvertebrate Sample
Project: North QLD Bloassessment Program
Site code: N@BP001
Site name:
Date: <u>22 / 03 / 2015</u> Time: <u>11:45</u> m/pm
Habitat sampled: <u>Edge</u> Preservative: <u>Ethanol</u>
Collector: <u>SKL</u> Picker: <u>GTH</u> Vial: <u>1</u> of <u>1</u>
Collection method: <u>10m sweep</u>
Picking time/method:

Figure 2: Example of a vial label for aquatic macroinvertebrate samples picked in the field

7.3.3 Curation and storage

- 1. When picking is completed, remove some of the liquid in the vial (which is usually diluted by water that has been added with the animals) using a mesh-covered syringe (Figure 3).
- 2. Refill the vial with fresh 70% ethanol or methylated spirits to the top and ensure the lid is tightly screwed on.
- 3. Ensure the vial you use is large enough. If the animals you collect take up more than 30% of the volume, use a larger container. Alternatively, use several vials clearly labelled to indicate that more than one vial was used e.g. 'vial 1 of 3'.
- 4. A label must be placed in the vial, an example of the kind of information that should accompany the

sample is presented in Figure 2. Label must be completed using pencil or alcohol-proof ink. Labels should also be printed in alcohol-proof ink.

5. Samples should be packed upright, in rigid lidded containers. Foam inserts are useful to ensure samples remain upright.



Figure 3: Mesh covered syringe

7.3.4 Quality assurance and quality control of live-picked aquatic macroinvertebrates

7.3.4.1 Field-based review of live picking

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

- 1. The sample is live picked by the operator (Section 7.3.2). During the pick, a field aquatic macroinvertebrate record sheet is filled in (Appendix 3).
- 2. An immediate re-pick is then undertaken by a second operator on the residue of the sample picked by the first operator. The second operator must be trained and accredited. The re-pick is undertaken in the same way as a normal field live-pick and the field aquatic macroinvertebrate record sheet completed by the first operator is also filled in by the second operator.
- 3. The field aquatic macroinvertebrate record sheet is immediately checked for inconsistencies, which are discussed between the two pickers to reduce systematic errors in subsequent live picks. If errors are large a check of a second sample should be considered.
- 4. Both samples are then preserved and transported to the laboratory for identification and further analysis.

7.3.4.2 Laboratory assessment of live pick

- 1. Samples from both operators are processed as outlined in Section 7.4.
- 2. Two statistical measures are then applied to the data:
 - The ratio of the live pick taxa richness from operator 1 to the total QA/QC sample taxa richness (sample from operator 1 + sample from operator 2) must be >0.90.
 - The Bray-Curtis Dissimilarity Index between samples picked by operator 1 and the total QA/QC sample taxa richness (sample from operator 1 + sample from operator 2) must be <0.2. Bray-Curtis Dissimilarity Index is based on presence/absence data.

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

7.4 Laboratory picking aquatic macroinvertebrates

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

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

- 3. Evenly distribute the remaining smaller fractions from the sieves into a subsampler. The subsampler used and recommended is a modified Marchant subsampler (Marchant, 1989) (Figure 4). This subsampler contains 100 circular cells, each 3.5cm in diameter x 3.5cm deep.
- 4. Fill the subsampler until the water level reaches the top of the cells, secure the lid, and rotate vigorously in both directions until the sample is distributed throughout the cells. Using a vacuum pump, subsample 10% (10 cells) of the whole sample, ensuring every 1% (1 cell) of the subsample is stored in separate containers. Label each container.
- 5. Sort and identify the subsamples (see sections 7.5.2 and 7.5.3 for a detailed method details), noting how many new taxa there are in every 1% subsample sorted.

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



Figure 4: Modified Marchant subsampler

7.5 Identification and enumeration of aquatic macroinvertebrates to family level

7.5.1 Sample preparation

- 1. Rinse the preservative from the sample by placing it in a 250µm sieve and gently running water through it, or use a plastic 250ml specimen jar with a mesh insert in the lid (Figure 5a).
- 2. Using a water squeeze bottle, flush the sieved contents into a large petri dish or sorting tray lid (Figure 5b). If the sample is large flush the sieve into a 250mL specimen jar. Small amounts of the sample and water can then be poured into a sorting tray or petri dish for processing. If there is excess water in the sample it can be removed using a 250µm mesh-covered syringe.

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

7.5.2 Sample processing

- 1. Place sorting tray under a dissecting microscope (minimum 10x magnification) and search systematically for aquatic macroinvertebrates.
- 2. Remove the animals from the sorting tray under the lowest magnification and place in water in small petri dishes or glass specimen blocks for later identification. Use a separate dish for each type of taxa. It may be desirable to separate the adults, larvae, and pupae of some taxa such as beetles (adults and larvae) and dipterans (larvae and pupae), for ease of identification.

- 3. Work your way through the sorting tray until all specimens have been removed.
- 4. Adjust the microscope to a higher magnification and pass over the sorting tray once more. This will ensure that the smaller specimens that may be missed at a lower magnification are seen.
- 5. Discard any non-macroinvertebrate material that may have been collected.
- 6. Select a polypropylene vial or glass jar that is large enough to fit the entire sample and a label. Use pencil or alcohol-proof ink to fill in details. The label must have all the information presented in Figure 6.
- 7. Place the label in the vial and half fill the vial with preservative (70% ethanol (or methylated spirits)/3% glycerol/27% water).
- 8. Select a group of specimens and identify each specimen using the appropriate taxonomic keys. Organisms are identified to various taxa level as outlined in the example Aquatic Macroinvertebrate Field Record Sheet presented in Appendix 3. If uncertain about the identity, obtain a second opinion from a colleague or local specialist. If a new taxon is suspected, contact a taxonomic specialist or alternatively the Queensland Museum.
- 9. When the specimen has been identified, place it in the vial and tally in the identification sheet (see example sheet provided in Appendix 4).
- 10. Once all taxa have been identified and placed in the vial, fill it with preservative (70% ethanol (or methylated spirits)/3% glycerol/27% water) and screw the lid on firmly.
- 11. Record the total tally for each taxon.







Identified Macroinvertebrate Sample									
Project: <u>North QLD Bioassessment Program</u>									
Site code: NQBP001									
Site name:									
Date: <u>22 / 03 / 2015</u> Time: <u>11:45</u> m)pm									
Habitat sampled: <u>Edge</u> Taxa: <u>All</u>									
Collector: <u>SKL</u> Picker: <u>GTH</u>									
ID by: <u>PRT</u> ID date: <u>01 / 04 / 2015</u>									
Vial <u>1</u> of <u>1</u> Preservative: <u>70% Ethanol</u>									

Figure 6: Example laboratory vial label

7.5.3 Quality assurance of taxonomic identification of aquatic macroinvertebrates to family level

7.5.3.1 QA/QC requirements

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

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

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

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

7.5.3.2 Calculation of quality assurance results

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

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

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

7.6 Long-term sample preservation and storage

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

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

7.7.1 Richness (taxa) index

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

7.7.2 Average SIGNAL index calculation

1. Allocate a SIGNAL grade number to each taxon in the sample. SIGNAL grade numbers for version 2.4 are available from the SIGNAL manual (Chessman 2003) which can be found at http://www.environment.gov.au/resource/signal-2iv-scoring-system-macroinvertebrates-water-bugs-

australian-rivers.

- 2. The SIGNAL Index is calculated for each sample by averaging the SIGNAL grade numbers of all of the aquatic macroinvertebrate taxa collected in a sample.
- 3. Taxa that do not have a SIGNAL grade number, for example Copepoda, Cladocera and Ostracoda, are not used in the calculation of the SIGNAL Index.

7.7.3 PET taxa richness

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

7.7.4 Calculating the % sensitive taxa index using presence/absence data

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

% Sensitive Taxa Index = Number of taxa with SIGNAL grade numbers ≥8 Total number of taxa in sample with SIGNAL grade numbers ×100

7.7.5 Calculating the % tolerant taxa index using presence/absence data

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

Number of taxa with SIGNAL grade numbers ≤ 3

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

8 References and additional reading

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Appendix 1 Equipment checklist

Table 1 Aquatic habitat identification

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

Table 2 Sampling aquatic macroinvertebrates using a dip net

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

Table 3 Live picking aquatic macroinvertebrates

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

Table 4 Identification of aquatic macroinvertebrates

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

Appendix 2 Velocity: Depth table

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

- V:D > 0.032 = riffle (R=Riffle in Table 5)
- V:D < 0.0124 = pool (P=Pool in Table 5)
- V:D 0.0124 0.032 = run (Run in Table 5)

			Velocity (m/sec)																								
		0	0.1	0.2	0.3	0.4	0.5	0.6	0.7	0.8	0.9	1.0	1.1	1.2	1.3	1.4	1.5	1.6	1.7	1.8	1.9	2.0	2.1	2.2	2.3	2.4	2.5
	5	Р	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
	10	Р	Р	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
	15	Р	Р	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
	20	P	P	P	run	run	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R	R
	25	, D	, D	D	D	run	rup	rup	run	rup	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P	P
	30		' D	' D	' D	run	run	run	run	run	TX FUE																
	35						Turi	Turi	Turi	Turi	Turi	ĸ	ĸ		R D											R D	
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~	45	P	Р	Р	Р	Р	run	run	R	R	R	R	R	R	R	R	R	R	R	R	R						
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ept	60	Р	Ρ	Р	Р	Ρ	Ρ	Р	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R	R	R	R
	00 65	Р	Ρ	Р	Р	Ρ	Ρ	Р	Ρ	run	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R	R
	65	Р	Р	Р	Р	Р	Ρ	Р	Ρ	Ρ	run	run	run	run	run	run	run	run	run	run	run	run	R	R	R	R	R
	70	Р	Ρ	Р	Р	Р	Ρ	Р	Ρ	Р	run	run	run	run	run	run	run	run	run	run	run	run	run	run	R	R	R
	75	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	R
	80	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
	85	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	run	run	run	run	run	run	run	run	run	run	run	run	run	run	run
	90	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	Р	run													
	95	P	P	P	P	P	P	P	P	P	P	P	P	run	run	run	rup	run	run	run							
	100	P	, Þ	' P	' P	P	' P	' P	' P	' P	' P	, Þ	, Þ	P	run												

Table 5 The Velocity: Depth table to identify stream habitat types (R = riffle and P = pool)

Appendix 3 Example aquatic macroinvertebrate field record sheet

(appears on following page)

Aquatic Macroinvertebrate Field Record Sheet									
Project:	Vial: of (e.g. 2 of 3)	Collectors initials:							
Site code:	Habitat: (e.g. Edge)								
Site name:	Picker 1 name:	Picker 2 name:							
Sampling run:	Picker 1 (original picker) signature:	Picker 2 (re-picker) signature:							
Sampling date: / / 20									

Picker 1 (original picker): tally number of specimens in column "1" next to the correct taxonomic group Picker 2 (re-picker): tally number of specimens in column "2" next to the correct taxonomic group

Lower Phyla Clacidotoldae s-T anypodinae Georisidae Zygoptera Rotifera Interidae Culcidae Gyrinidae Retigeobarcesia Interidae Portifora Umanaidae Dolichopodidae Halipidae Calopterygidae Interidae Openifora Phynsidae Empiddae Halipidae Cohorismagrionidae Interidae Openifora Phynoidae Empiddae Hydrochidae Diphebidae Interidae Openifora Phynoidae Ephydridae Hydrochidae Diphebidae Interidae Openifora Phanobidae Ephydridae Hydrophildae Interidae Interidae Openifora Phanobidae Ephydridae Hydrophildae Interidae Interidae Nematoda Corbiculdae Synteeridae Siniulidae Nanophydridae Interidae Oligochaeta Megaloptera Tabanidae Nanophydridae Aesthridae Interidae Hurdinae Ephorecoptera Tabanidae Scritidae Aesthridae Interidae		1	2		1	2		1	2		1	2		1	2
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CopepodaOsmylidaeLeptopodidaeDipseudopsidaeUrothemistidaeINotostracaSisyridaeMesoveliidaeEcnomidaeMothsIPsammaspididaePlecopteraMicronectidaeGlossosomatidaeCrambidaeIAmphi-ChiltoniidaeAustroperlidaeNaucoridaeHelicophidaeICrambidaeIAmphi-CorophiidaeEustheniidaeNaucoridaeHelicopsychidaeIIIAmphi-CorophiidaeGripopterygidaeNotonectidaeHelicopsychidaeIIIAmphi-EusiridaeGripopterygidaeNotonectidaeHydropsychidaeIIIIso-SphaeromatidaeNotonemouridaeOchteridaeHydropsychidaeIIIAtyidaeAthericidaeSaldidaeILeptoceridaeIIIPalaemonidaeCeratopogonidaeVeliidaeIPhiloptarnidaeIIIParastacidaeCorethrellidaeCranbidaePhiloptarnidaeIIIIParathelphusidaeChironomidaeCurculionidaePolycentropodidaeIIIIAncylidaeS-f ChironominaeDytiscidaePsychomylidaeIIIIMoluscaS-f OrthocladiinaeElmidaeStenopsychidaeIIIIParathelphusidaeS-f ChironominaeICurculionidaePsychomylidaeIIIIParathelphusidaeS-f ChironominaeIDy	Ostracoda			Neurorthidae			Hydrometridae			Conoesucidae			Telephlebiidae		
NotostracaSisyridaeMesoveliidaeEcnomidaeMothsIPsammaspididaeIPlecopteraIMicronectidaeIGlossosomatidaeICrambidae ex-Pyralidae]IAmphi-ChiltoniidaeIAustroperlidaeINaucoridaeIHelicophidaeIIAmphi-CorophiidaeIEustheniidaeINepidaeIHelicopsychidaeIIAmphi-CorophiidaeIEustheniidaeINotonectidaeIHydrobiosidaeIIAmphi-EusiridaeISripopterygidaeINotonectidaeIHydropsychidaeIIIso-SphaeromatidaeINotonemouridaeIOchteridaeIHydropsychidaeIIIso-CorallanidaeIDipteraISaldidaeIHydropsychidaeIIIPalaemonidaeISaldidaeIIOdontoceridaeIIIIParastacidaeICorethrellidaeIIPolycentropodidaeIIIIParathelphusidaeIChaoboridaeIChrysomelidaeIPolycentropodidaeIIIIMolluscaIChironominaeIDytiscidaeIPsychomyidaeIIIIIParathelphusidaeIS-f ChironominaeIDytiscidaeIPsychomyidaeIIIIIIIParathelphusidae	Copepoda			Osmylidae			Leptopodidae			Dipseudopsidae			Urothemistidae		
PsammaspididaePlecopteraMicronectidaeGlossosomatidaeCrambidae (ex-Pyraildae)Crambidae (ex-Pyraildae)Crambidae (ex-Pyraildae)Image: CombidaeImage: Combidae (ex-Pyraildae)Image: Combidae 	Notostraca			Sisyridae			Mesoveliidae			Ecnomidae			Moths		
Amphi-Chiltoniidae [Ex. Ceinidae]AustroperlidaeNaucoridaeHelicophidaeImage: Ceinidae]Amphi-CorophiidaeEustheniidaeNepidaeHelicopsychidaeOTHER TAXAImage: Ceinidae]Amphi-EusiridaeGripopterygidaeNotonectidaeHydrobiosidaeImage: Ceinidae]Image: Ceinidae]Image: Ceinidae]Iso-SphaeromatidaeNotonemouridaeOchteridaeHydropsychidaeImage: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]Iso-CorallanidaeDipteraPleidaeHydropsychidaeImage: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]AtyidaeAthericidaeSaldidaeLeptoceridaeImage: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]ParastacidaeCeinetopogonidaeColeopteraPhilopotamidaeImage: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]Image: Ceinidae]ParastacidaeCorethrellidaeCeinonomidaeCeinonomidaeImage: Ceinidae]Image: Ceinidae]	Psammaspididae			Plecoptera			Micronectidae			Glossosomatidae			Crambidae [ex-Pyralidae]		
Amphi-CorophildaeEustheniidaeNepidaeHelicopsychidaeOTHER TAXAImage: Constraint of the system of	Amphi-Chiltoniidae			Austroperlidae			Naucoridae			Helicophidae					
Amphi-EusiridaeIGripopterygidaeNotonectidaeHydrobiosidaeIIIIIso-SphaeromatidaeNotonemouridaeOchteridaeHydropsychidaeIIIIIso-CorallanidaeDipteraPleidaeHydroptilidaeIIIIAtyidaeAthericidaeSaldidaeILeptoceridaeIIIPalaemonidaeBlephaceridaeVeliidaeIOdontoceridaeIIIParastacidaeCeratopogonidaeVeliidaePhilopotamidaeIIIIParathelphusidaeCorethrellidaeIChrysomelidaePolycentropodidaeIIITrogloplacidaeChironomidaeICurculionidaePsychomyiidaeIIIIAncylidaeS-f ChironominaeIDytiscidaeITasimiidaeIIIIBithyniidaeS-f OrthocladiinaeSelmidaeIStenopsychidaeIIIIBithyniidaeISelmidaeIIIIIIIItageIIIIIIIIIItageIIIIIIIIIIItageIIIIIIIIIIItageIIIIIIIIIIItageII </td <td>Amphi-Corophiidae</td> <td></td> <td></td> <td>Eustheniidae</td> <td></td> <td></td> <td>Nepidae</td> <td></td> <td></td> <td>Helicopsychidae</td> <td></td> <td></td> <td>OTHER TAXA</td> <td></td> <td></td>	Amphi-Corophiidae			Eustheniidae			Nepidae			Helicopsychidae			OTHER TAXA		
Iso-SphaeromatidaeNotonemouridaeOchteridaeHydropsychidaeIIIIso-CorallanidaeDipteraPleidaeHydroptilidaeIIIIAtyidaeAthericidaeSaldidaeLeptoceridaeIIIIPalaemonidaeBlephaceridaeVeliidaeIOdontoceridaeIIIIParastacidaeCeratopogonidaeVeliidaeIPhilopotamidaeIIIIParasthelphusidaeCorethrellidaeICarabidaePhiloprterithridaeIIIITrogloplacidaeChironomidaeICurculionidaePolycentropodidaeIIIIAncylidaeS-f ChironominaeIDytiscidaeITasimiidaeIIIIBithyniidaeS-f OrthocladiinaeIElmidaeStenopsychidaeIIII	Amphi-Eusiridae			Gripopterygidae			Notonectidae			Hydrobiosidae					
Iso-CorallanidaeDipteraPleidaeHydroptilidaeImage: SaldidaeHydroptilidaeImage: SaldidaeHydroptilidaeImage: SaldidaeImage: Sald	Iso-Sphaeromatidae			Notonemouridae			Ochteridae			Hydropsychidae					
AtyidaeAthericidaeSaldidaeLeptoceridaeIAthericidaeIPalaemonidaeBlephaceridaeVeliidaeOdontoceridaeIIIParastacidaeCeratopogonidaeColeopteraPhilopotamidaeIIIParathelphusidaeCorethrellidaeCarabidaePhiloprhreithridaeIIITrogloplacidaeChaoboridaeChrysomelidaePolycentropodidaeIIIMolluscaChironomidaeCurculionidaePsychomyiidaeIIIAncylidae (part Planotbidae]S-f ChironominaeDytiscidaeITasimiidaeIIBithyniidaeS-f OrthocladiinaeElmidaeStenopsychidaeIIII	Iso-Corallanidae			Diptera			Pleidae			Hydroptilidae					
PalaemonidaeImage: BlephaceridaeVeliidaeImage: Delta controlOdontoceridaeImage: Delta controlImage: Delta control <t< td=""><td>Atyidae</td><td></td><td></td><td>Athericidae</td><td></td><td></td><td>Saldidae</td><td></td><td></td><td>Leptoceridae</td><td></td><td></td><td></td><td></td><td></td></t<>	Atyidae			Athericidae			Saldidae			Leptoceridae					
ParastacidaeCeratopogonidaeColeopteraPhilopotamidaeImage: Construction of the state	Palaemonidae			Blephaceridae			Veliidae			Odontoceridae					
ParathelphusidaeCorethrellidaeCarabidaePhilorhreithridaeImage: CorethrellidaeImage: CorethrellidaeTrogloplacidaeChaoboridaeChrysomelidaePolycentropodidaeImage: CorethrellidaeImage: C	Parastacidae			Ceratopogonidae			Coleoptera			Philopotamidae					
TrogloplacidaeChaoboridaeChrysomelidaePolycentropodidaeImage: ChironomidaeMolluscaChironomidaeCurculionidaePsychomylidaeImage: ChironomidaeImage: ChironomidaeAncylidae part Planobidae]s-f ChironominaeDytiscidaeTasimiidaeImage: ChironomidaeImage: ChironomidaeBithyniidaes-f OrthocladiinaeElmidaeStenopsychidaeImage: ChironomidaeImage: Chironomidae	Parathelphusidae			Corethrellidae			Carabidae			Philorhreithridae					
MolluscaChironomidaeCurculionidaePsychomyiidaeImage: ChironomidaeImage: ChironomidaeAncylidae (part Planorbidae)s-f ChironominaeDytiscidaeTasimiidaeImage: ChironomidaeImage: ChironomidaeBithyniidaes-f OrthocladiinaeElmidaeStenopsychidaeImage: ChironomidaeImage: Chironomidae	Trogloplacidae			Chaoboridae			Chrysomelidae			Polycentropodidae					
Ancylidae s-f Chironominae Dytiscidae Tasimiidae Image: Chironominae Image: Chironomi	Mollusca			Chironomidae			Curculionidae			Psychomyiidae					
Bithyniidae s-f Orthocladiinae Elmidae Stenopsychidae	Ancylidae			s-f Chironominae			Dytiscidae			Tasimiidae					
	Bithyniidae			s-f Orthocladiinae			Elmidae			Stenopsychidae					

Appendix 4 Example of a laboratory identification sheet (appears on following page)

Aquatic Macroinvertebrate Identification Sheet

Project:		
Site code:	Vial: of (e.g. 2 of 3)	I.D. by:
Site name:	Habitat: (e.g. Edge)	I.D. date:// 20
Sampling run:	Collectors initials:	Entered by:
Sampling date: / / 20	Picker initials:	Entry checked by:

Counts by taxa

Lower Phyla	Glacidorbidae	s-f Tanypodinae	Georissidae	Zygoptera	
Rotifera	Tateidae [ex-Hydrobiidae]	Culicidae	Gyrinidae		
Porifera	Lymnaeidae	Dixidae	Haliplidae	Calopterygidae	
Hydridae	Neritidae	Dolichopodidae	Heteroceridae	Chorismagrionidae	
Clavidae	Physidae	Empididae	Hydraenidae	Coenagrionidae	
Dugesiidae	Planorbidae	Ephydridae	Hydrochidae	Diphlebiidae	
Temnocephaloidea	Thiaridae	Muscidae	Hydrophilidae	Isostictidae	
Nemertea	Viviparidae	Psychodidae	Hygrobiidae	Lestidae	
Nematoda	Corbiculidae	Sciomyzidae	Limnichidae		
Gordiidae	Hyriidae	Simuliidae	Nanophyidae	Synlestidae	
Oligochaeta	Sphaeriidae	Stratiomyidae	Noteridae	Epiprocta	
Polychaeta	Megaloptera	Tabanidae	Psephenidae	Aeshnidae	
Hirudinea	Corydalidae	Tanyderidae	Ptilodactylidae	Austrocorduliidae	-
Glossiphoniidae	Sialidae	Thaumaleidae	Scirtidae	Cordulephyidae	
Hirudinidae	Ephemeroptera	Tipulidae	Spercheidae	Corduliidae	
Erpobdellidae	Ameletopsidae	Hemiptera	Sphaeriusidae	Gomphidae	
Acarina	Baetidae	Aphelocheiridae	Staphylinidae	Hemicorduliidae	
Collembola	Caenidae	Belostomatidae	Trichoptera	Libellulidae	
Sminthurididae	Ephemerellidae	Corixidae	Antipodeciidae	Lindeniidae	
Crustacea	Leptophlebiidae	Gelastocoridae	Atriplectididae	Macromiidae	
Spinicaudata	Prosopistomatidae	Gerridae	Calamoceratidae	Pseudocorduliidae	
Cladocera	Neuroptera	Hebridae	Calocidae	Synthemistidae	
Ostracoda	Neurorthidae	Hydrometridae	Conoesucidae	Telephlebiidae	
Copepoda	Osmylidae	Leptopodidae	Dipseudopsidae	Urothemistidae	
Notostraca	Sisyridae	Mesoveliidae	Ecnomidae	Moths	
Psammaspididae	Plecoptera	Micronectidae	Glossosomatidae	Crambidae [ex-Pyralidae]	
Amphi-Chiltoniidae	Austroperlidae	Naucoridae	Helicophidae		
Amphi-Corophiidae	Eustheniidae	Nepidae	Helicopsychidae	OTHER TAXA	
Amphi-Eusiridae	Gripopterygidae	Notonectidae	Hydrobiosidae		
Iso-Sphaeromatidae	Notonemouridae	Ochteridae	Hydropsychidae		
Iso-Corallanidae	Diptera	Pleidae	Hydroptilidae		
Atyidae	Athericidae	Saldidae	Leptoceridae		
Palaemonidae	Blephaceridae	Veliidae	Odontoceridae		
Parastacidae	Ceratopogonidae	Coleoptera	Philopotamidae		
Parathelphusidae	Corethrellidae	Carabidae	Philorhreithridae		
Trogloplacidae	Chaoboridae	Chrysomelidae	Polycentropodidae		
Mollusca	Chironomidae	Curculionidae	Psychomyiidae		
Ancylidae [part anorbidae])	s-f Chironominae	Dytiscidae	Tasimiidae		
Bithyniidae	s-f Orthocladiinae	Elmidae	Stenopsychidae		

NOTES [e.g. taxa kept aside/sent away for expert ID]:

Appendix 5 Example of a QA/QC laboratory identification sheet

(appears on following page)

Project:		Vial: of	(e.g. 2 of 3)	QA/QC date:// 20
Site code:		Habitat:	(e.g. Edge)	QA/QC I.D. by:
Site name:		Picker initials:	\	/erification by:
Sampling run:		Original I.D. by:	E	Entered by:
Sampling date:	/ / 20	I.D. date: /	/ 20 E	Entry checked by:
Counts by taxa o	A Q	A Q	A	
ower Phyla	Glacidorbidae	s-f Tanypodinae	Georissidae	Zygoptera
Rotifera	Tateidae	Culicidae	Gyrinidae	Argiolestidae
orifera	Lvmnaeidae	Dixidae	Haliplidae	
lydridae	Neritidae	Dolichopodidae	Heteroceridae	Chorismagrionidae
Javidae	Physidae	Empididae	Hydraenidae	Coenagrionidae
Dugesiidae	Planorbidae	Ephydridae	Hydrochidae	Diphlebiidae
emnocephaloidea	Thiaridae	Muscidae	Hydrophilidae	Isostictidae
lemertea	Viviparidae	Psychodidae	Hygrobiidae	Lestidae
lematoda	Corbiculidae	Sciomyzidae	Limnichidae	Platycnemidae
Gordiidae	Hyriidae	Simuliidae	Nanophyidae	Synlestidae
Dligochaeta	Sphaeriidae	Stratiomvidae	ex-Brentidae]	Epiprocta
Polychaeta	Megaloptera	Tabanidae	Psephenidae	Aeshnidae
lirudinea	Corvdalidae	Tanyderidae	[adults terrestrial]	Austrocorduliidae
Glossiphoniidae	Sialidae	Thaumaleidae	Scirtidae	
lirudinidae	Ephemeroptera	Tipulidae	Spercheidae	Corduliidae
rpobdellidae	Ameletopsidae	Hemiptera	Sphaeriusidae	Gomphidae
carina	Baetidae	Anhelocheiridae	[ex-Microsporidae]	Hemicorduliidae
Collembola	Caenidae	Belostomatidae	Trichoptera	
	Ephemerellidae	Corixidae	Antipodeciidae	
Crustacea	Leptophlebiidae	Gelastocoridae	Atriplectididae	Macromiidae
Spinicaudata	Prosopistomatidae	Gerridae	Calamoceratidae	Pseudocorduliidae
ladocera	Neuroptera	Hebridae	Calocidae	Synthemistidae
Dstracoda	Neurorthidae	Hydrometridae	Conoesucidae	Telephlebiidae
Copepoda	Osmylidae	Leptopodidae	Dipseudopsidae	Urothemistidae
lotostraca	Sisyridae	Mesoveliidae	Ecnomidae	Moths
sammaspididae	Plecoptera	Micronectidae	Glossosomatidae	
mphi-Chiltoniidae	Austroperlidae	Naucoridae	Helicophidae	
Amphi-Corophiidae	Eustheniidae	Nepidae	Helicopsychidae	OTHER TAXA
mphi-Eusiridae	Gripopterygidae	Notonectidae	Hydrobiosidae	
so-Sphaeromatidae	Notonemouridae	Ochteridae	Hydropsychidae	
so-Corallanidae	Diptera	Pleidae	Hydroptilidae	
tyidae	Athericidae	Saldidae	Leptoceridae	
alaemonidae	Blephaceridae	Veliidae	Odontoceridae	
arastacidae	Ceratopogonidae	Coleoptera	Philopotamidae	
'arathelphusidae	Corethrellidae	Carabidae	Philorhreithridae	
rogloplacidae	Chaoboridae	Chrysomelidae	Polycentropodida	ie
/lollusca	Chironomidae	Curculionidae	Psychomyiidae	
ncylidae [part lanorbidae]	s-f Chironominae	Dytiscidae	Tasimiidae	
Bithvniidae	s-f Orthocladiinae	Elmidae	Stenopsvchidae	