FZP Protocol

CRUSTACEAN ZOOPLANKTON

Aim

To monitor changes in the abundance of crustacean zooplankton in standing waters.

Rationale

The crustacean zooplankton which dominate the open water of lakes form an important link in the food chain between phytoplankton and fish. Most lakes contain a few species from a small number of genera but marked changes in species composition can occur when there is a progressive change in the trophic status. The distribution and occurrence of freshwater zooplankton populations are notoriously patchy. Different species tend to concentrate at different depths and there are often local accumulations produced by wind-induced water movements (George 1981). Detailed population studies require very intensive sampling but a single integrated sample collected from a central location can produce useful results for more general surveys of seasonal and inter-annual change.

Method

Equipment

Samples are collected with a net fitted with a non-porous reducing cone to increase the efficiency of filtration and designed according to the principles outlined in Tranter and Smith (1968). The area of gauze used in the net must be large enough to filter the water efficiently and the porosity of the gauze chosen to minimise the risk of clogging. The type of mesh used for the construction of a net has a marked effect on the efficiency and selectivity of the net. Nets made from stainless steel mesh are robust, easy to clean and have more precise mesh dimensions, but nets made from nylon are more readily obtainable from commercial suppliers; nets with a diameter of 25 cm will normally be used. Nets with a mesh size of 140–150 µm, which deliberately exclude rotifers, are recommended (see Appendix I, page 89). The area and mesh size of nets used at any site must in any case be standardised and reported to the CCU.

Small glass, polyethylene or polycarbonate containers can be used to store zooplankton samples. The containers selected should be reasonably robust and fitted with lids or closures which minimise the quantity of liquid lost by evaporation during prolonged storage.

Location

Samples should be collected preferably fortnightly, but not less than quarterly, at a central location near the deepest point in the lake. In circumstances where a boat is not available for sampling it is permissible to sample zooplankton at the lake outflow or from a jetty or dam which projects over deep water.

The grid reference of the sampling site should be recorded as accurately as possible, usually to within 30–40 m, together with the water depth at the time of sampling.

Sampling

Samples are collected by hauling a net, at a steady speed, from the bottom to the surface of the standing water. Where water depth exceeds 100 m the net should be hauled from a depth of 100 m to the surface. Care should be taken to wash any animals adhering to the side of the net into the collecting vessel. The net should be washed in lake water after use and then dried to minimise the risk of transferring organisms between sites. Samples should be killed in the field by adding a few drops of 40% formaldehyde to the sample in its storage container. If too much formaldehyde is added in the field, any Cladocera in the sample will shed a high proportion of their eggs which may then be lost during subsequent laboratory processing.

In-house procedures for using formaldehyde are provided by the Environment Agency (1997). These procedures have been tested to meet statutory safety standards (COSHH); however, every laboratory using these procedures, or other procedures authorised by their own laboratory, must carry out its own safety assessment, tailored to its own particular conditions and facilities. 100 ml of undiluted formalin (40% aqueous formaldehyde) should be placed in a 150 ml Nalgene screw-topped bottle in the laboratory, using a fume cupboard or fume extractor. A bottle of fixative should be placed in each sample container. In the field a few drops of the formalin should be added to the sample. The cap is then replaced on the bottle containing the unused formalin this is itself replaced in the sample container. This procedure must take place outside, in a well-ventilated area and **not** inside a vehicle. Adherence to this procedure ensures that the formalin is always double-sealed, prevents large volumes of fixative being carried in the same container and limits the total volume being carried to that required for sampling. Protective gloves must always be worn when handling concentrated formalin.

Samples may be stored either in formaldehyde or in alcohol. If the samples are to be stored in formaldehyde, enough 40% formaldehyde should be added on returning to the laboratory to give a final concentration of 4–5%. If the samples are to be preserved in alcohol, the animals should be removed by filtering through a suitable mesh and then re-suspended in 70% alcohol. When using either formaldehyde or alcohol, precautions must be taken to minimise skin contact and avoid exposing operators to vapour. Samples should be washed with water before sorting and re-suspended in preservative as soon as they have been examined microscopically.

Counting procedures

Several types of counting chamber have been designed for examining zooplankton in the laboratory. The simplest are machined from a rectangular block of 'Perspex' but the most convenient are those designed to rotate on the stage of a stereo-zoom microscope. Jones (1979) describes and illustrates the type of circular 'Perspex' moat used at the IFE, Windermere, for zooplankton counting. The sample is dispensed into a machined 'moat' fitted with a radial barrier and the chamber is rotated on its central spindle until all the animals have been counted. If a sub-sampling procedure is used to prepare material for counting, its nature must be reported to the CCU. Counts are reported as number of individuals per litre by species.

Estimation of biomass

An estimate of zooplankton biomass is a useful additional measure. Acceptable estimates of the seasonal and inter-annual changes in biomass can be obtained using simple volumetric techniques. George and White (1985) compared a rapid method of estimating the settled volume of preserved plankton with a more complex displacement volume method (Tranter 1959). The results demonstrated that reliable settled volume estimates can be obtained for most species of freshwater zooplankton, but the glass tubes used for such measurements should be carefully matched to the expected volume of sample. Samples are normally stored in flat-bottomed glass tubes with an internal diameter of 1.8 cm and the height of the settled material is measured with a transparent ruler.

Settled volume is reported as millilitres (ml).

Archiving samples

Zooplankton samples should be archived by storing in 4% formaldehyde containing a small quantity (2–3%) of glycerol to protect the samples from evaporation. Samples should be checked periodically for evaporative loss.

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References

Environment Agency 1997. *Procedures for collecting and analysing macro-invertebrate samples.* Environmental Monitoring Programme: Biological Techniques, BT001. (Internal Publication), Bristol: Environment Agency.

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Tranter, D.J. 1959. A method for determining zooplankton volumes. *Journal du Conseil*, **25**, 272–278.

Tranter, D.J. & Smith, P.E. 1968. Filtration performance. In: *Zooplankton sampling. Monographs on oceanographic methodology 2*. Paris: UNESCO.

Appendix I

Nets of a suitable quality and specification are obtainable from various biological equipment suppliers for approximately £50. Details of suppliers can be obtained from the CCU.

Specification of results and recording conventions

The measurement variables listed below are those required for each FZP sampling location at an ECN Site. Sites submitting data to the ECNCCU should refer to the accompanying Data Transfer documentation for the specification of ECN dataset formats, available on the restricted access Site Managers' extranet. Contact ecnccu@ceh.ac.uk if you need access to this documentation.

The first 4 key parameters uniquely identify a sample or recording occasion in space and time, and must be included within all datasets:

• Site Identification Code (e.g. R10)	Unique code for each ECN Site
• Core Measurement Code (e.g. FWC)	Unique code for each ECN 'core measurement'
• Location Code (e.g. 01)	Each ECN Site allocates its own code to replicate sampling locations for each core measurement (e.g. FWC 01, FWC 02 for different surface water collection points)
• Sampling Date (/time)	Date on which sample was collected or data recorded. This will include a time element where sampling is more frequent than daily

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Core measurement: Crustacean zooplankton (FZP protocol)

The following variables are recorded from samples taken at a recommended frequency of fortnightly, at lake sites only:

Variable	Units	Precision of recording
Site identification code		
Core measurement code		
Location code		
Sampling date		
Sampling time	GMT (24h)	1 min
Net mesh size	µm	1
Net bag depth	mm	1
Net mouth area	mm^2	1
Settled volume	ml	1
For each species present:		
Species code	8-digit code(1)	(eg 31130201)
Species name	genus species	(eg *Eudiaptomus gracilis*)
Concentration	indivs 1^{-1}	

Note

(1) ECN uses the 'revised Maitland' coding system (Furse *et al*, 1989) used by RIVPACs. A list of crustacean zooplankton, which forms a sub-set of the RIVPACs list, has been provided by the Institute of Freshwater Ecology; a copy of this species list and associated RIVPACs codes can be obtained from the CCU. A copy of the complete RIVPACs coding system may be obtained through the CCU.