Technical reference material: collection, processing and preservation of marine biota for tissue burden contaminant analysis

Operational instruction **Issued** 11/10/07 **Review due by** 11/01/09

Contents

Document details

Related documents

Feedback

Contact for queries

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1.0 Purpose

This document is aimed at all staff involved in collecting and processing marine (estuarine and coastal) biota samples for chemical analysis – i.e. bioaccumulation studies. It provides information on the factors that must be considered when designing a sampling programme to look at bioaccumulation; and how to collect, process and preserve samples prior to their analysis by the National Laboratory Service.

2.0 Competency

2.1 Health and safety

The collection of samples for bioaccumulation can involve working on soft and hard substrata and from a boat, therefore all fieldworkers should be familiar with the relevant Health & Safety documentation:

- 27_04 GRA - Field work, 725_06 Fieldwork
- 600_06 Lifejackets and buoyancy aids
- 426_05 A - Working in or near water, 732_06 Working in or near water
- 193_06 Intertidal Soft Sediment, 13_07 Intertidal Soft Sediment

- 32_04 GRA Boatwork, 730_06 Working from a boat
- e-Learning - Understanding Tides and Tide Tables

In the laboratory, laboratory protocols must be followed:

- 242_06 GRA – Ecology Laboratory Safety, 52_05 Ecology Laboratory Safety

2.2 Collection and dissection

- General training and assessment for this work is achieved through completing the Marine ecology monitoring CDrom and eLearning on-line assessment.
- Staff must have read the General sampling procedures, which provides good sample handling proactive for chemical analysis
- For training in dissection, competency can be achieved by In-house training by a suitably competent person and completing the Cdrom.
- OR for fish dissection only, Applied Fish Health Foundation Course run by the National Fisheries
  Laboratory, (the course includes some tuition on fish dissection) – are suitable

In addition to the above, further guidance on dissection can be found in:

- The Quest for Quality Sample Handling – a Marine Laboratory (Aberdeen) video. Produced as part of the Quality Assurance of Information for Marine Environmental Monitoring in Europe (QUASIMEME) Project (contact ISU) concentrating on fish processing.

3.0 Introduction

The monitoring of contaminant levels in coastal and estuarine species is carried out for the following reasons:

- to provide information on spatial and temporal trends of contaminants in the environment.
- to monitor changes following programmes to reduce inputs of chemicals and metals to the environment
- to investigate specific biological effects and their chemical cause

Chemical analysis on biological tissue offers several benefits for monitoring contaminants in the marine environment:

- contaminants in biological tissue are an indication of their persistence in the environment
- the levels of contaminants in tissue reflect the biologically available levels
- biomagnification of contaminants provides concentrations amenable to readily available analysis techniques

These advantages have led to widespread adoption of bioaccummulation work in programmes such as CSEMP (originally NMMP) and monitoring for the Titanium Dioxide Directive.

4.0 Target species

An appropriate species must be selected for sampling. There is no one species that is capable of acting as a universal “indicator”. Many species are inappropriate because they are scarce, difficult to handle, or because they regulate the body-burden of contaminants in some way.

There are a variety of ways in which contaminants are accessible to biota. I.e. through dissolved contaminants; contaminants associated with sediments, detritus and particulates; contaminants present in organisms consumed by other organisms.
Different species will take up these contaminants in different ways, depending on their physiology and feeding mechanisms. Table 1. details species and the ways (also known as phase) in which contaminants are taken up by each.

Table 1. List of species suitable for coastal and estuarine bioaccumulation studies, and the environmental compartment of which they are indicative (After Barnett, 1990).

Target species	**Environmental compartment (phase)**
Fucus vesiculosus (Bladder wrack) Fucoid algae (preferably *Fucus serratus*)	Dissolved constituents
Mytilus edulis (Mussel) *Ostrea edulis* (Native oyster) *Cerastoderma edule* (Cockle)	Dissolved and particulate persistent contaminants
Patella vulgata (Limpet) *Littorina littorea* (Common Winkle)	Dissolved and detrital constituents
Nereis (Hedistes) diversicolor (Ragworm)	Persistent contaminants present in sediments
Limanda limanda (Dab) *Platichthys flesus* (Flounder) *Pleuronectes platessa* (Plaice) *Merlanguis merlangus* (Whiting)	Dissolved constituents and dietary sources

In general, a comprehensive sampling programme should plan to include a variety of species, to cover the uptake of dissolved, detrital, particulate, sedimentary and dietary contaminants. Or, sampling can be tailored to answer specific questions. Table 2. summarises the determinands that can be monitored for in the different species.

The choice of certain species including Flounder and dab will provide consistency with existing international monitoring commitments.

When target species are chosen, the following factors must be taken into account when planning collection for bioaccumulation analysis:

- time of year with regard to the reproductive state of the organism (NB: this may vary geographically due to seasonal patterns). Collection must be outside of spawning periods.
- time of year with regard to physiological variations (e.g. intermoult stages in shrimp)
- size / weight / age
- tidal height of collection
- the number of specimens required in a sample to minimise natural variation

For temporal studies it is important that the species; time of year for sampling; tidal height; size range,weight ,age, physiological and sample size factors remain constant year on year.

Table 2.

A summary of when and where species should be collected, as well as the size range and number of specimens required is given in Table 3.

Summary of determinands monitored using various target species (After Barnett, 1990).

Target Species	Determinands efficiently accumulated/monitored	Determinands NOT reliably monitored	Biological determinations
F.vesiculosus (Fucoid algae)	As Cd CO Cu Ni Zn (Moderate efficiency for HG Se Sn)	Cr Pb	Dry wt. General condition
Limanda limanda (Dab)*)*	Hg from white muscle, Organic compounds, [Pesticides, PCB's, etc] and Cd from liver tissue)	Most metals	Sample wet wt. Length/length range, Lipid content (essential for assessing PCBs)
Platichthys flesus (Flounder)	As above	As above	As above
Pleuronectes platessa (Plaice)	As above	As above	As above
Gadus morhua (cod)	As above	As above	As above
Merlangius merlangus (Whiting)	As above	As above	As above
Mytilus edulis (Mussels)	Cd Cr Pb (Moderate efficiency for Co Hg Se Sn)	Cu, Zn, As, Ag	Wet wt. + shell Dry wt. - shell
Ostrea edulis (Oysters)	Cu Zn and most other metals		Wet wt. + shell Dry wt. - shell
Cerastoderma edule (Cockles)	Moderate efficiency -Ag As Ni Cd	Cu, Zn	Wet wt. + shell Dry wt. - shell
Patella vulgata (Limpet)	Cd Cu Moderate efficiency - Ag Hg Pb	As	Wet wt. + shell Dry wt. - shell
Littorina littorea (Winkle)	Ag Cd Pb (Moderate efficiency for As Hg)	Co Ni , Cr Mn Fe Zn	Wet wt. + shell Dry wt. - shell
Crangon crangon (Brown Shrimp)	Hg	Shrimp do not bioaccumulate PCBs Zn, Cu, Fe and Cd affected by intermoult cycle	

Table 3. Principle sample design criteria for target species used in bioaccumulation studies.

Target Species	No. of specimens in each batch	Size (range) mm	Time/Season for collection	Tidal/Shore position	Note
Fucus vesiculosus (Bladder wrack)	30-35	250-300	Feb-Mar to avoid growth and reproductive cycles	Mid-shore	
Fucoid algae	30-35	200-300	Feb-Mar	Mid-shore	*Fucus serratus* as first choice alternative
Platichthys flesus (Flounder)	30 analysed individually or in batches of 5	150-300	Outside spawning i.e.Sept-Dec	Shallow areas	Size range 250-300mm for North Sea. Period may be earlier depending on location see JAMP guidelines
Limanda limanda (Dab)	30 analysed individually or in batches of 5	200-300	Outside spawning i.e late Jun-Jan	Shallow areas	Size range 200-250mm for North Sea
Mytilus edulis (Mussels)	60	30-60	Mid.Jan-mid.Mar (i.e. before spawning)	Mid-shore	Size range and number per batch based on JAMP guidelines
Ostrea edulis (Oysters)	25-30	60-100 NB: min size may be dependent on local fishing regulations and must be set in consultation with the appropriate seafish authority	Jan-Mar. This may be influenced by local fishing seasons, and consult sea fisheries authority	Shallow sub-tidal	
Cerastoderma edule (Cockles)	50	25-40	Jan-Mar	Mid-shore	
Patella vulgata (Limpet)	50	40 - (measured across the greatest width)	Mar-May	Mid-shore	
Littorina littorea (Winkle)	30	ca. 20	Aug-Nov	Mid-shore	
Nereis diversicolor (Ragworm)	100	> 50 < 200	Aug/Sept However, Populations can have a secondary spawning season. Local knowledge required to determine the ideal sampling period	Mid-shore	There is uncertainty over appropriate batch size, this is an estimate.
Crangon crangon (Brown shrimp)	100-250	50-70 (rostrum to telson)	Avoid moult cycle	N/A	

5.0 Collection, processing and preservation

When collecting species, appropriate field observations should be recorded to provide information on visual evidence relating to possible contamination.

For fish, note the presence, prevalence and position of any evidence of diseases in the initial catch (e.g. fin-rot, carcinomas, ulcers and lymphocystis), do not confuse these with damage inflicted during trawling (see Appendix 3 & 4 of the Technical Reference Material document - 011_07 Trawling for fish (otter, beam & oyster dredge). Damaged fish or fish in poor condition must not be selected (NPMMG, 1999).

All samples for bioaccumulation analysis should be kept cool during transport to the laboratory, samples should be in clearly labelled plastic bags or clean polythene bucket with seal-tight lid.

Table 4 details the methods of transport, storage, cleansing, depuration, and target tissue for each species.

Table 4. Summary of tissue preparation, procedures and requirements (After Barnett, 1990).

Target Species	Transport following collection to the laboratory	Storage prior to cleansing/preparation	Cleansing	Depuration	Storage prior to dissection	Tissue Selection	Storage prior to analyses	Reference for further details
F.vesiculosus (and other *Fucus* spp.)	Keep cool, carry in a clean food-grade plastic bag	Refrigeration (up to 14 days)	Scrub with nylon brush and rinse with clean water. Scrape off epiphytes & dirt	N/A	No	Old thallus only	Freeze or analyse immediately	Barnett *et al* (1989) Bryan *et al* (1980)
Flatfish *(L.limanda) (P.flessus)*	Keep cool, return to laboratory ASAP	Refrigeration (up to 24 hrs only)	Thorough washing and gentle scrubbing, remove mucilage and attached matter	N/A	Deep frozen (for up to 12 mths)	Remove white muscle tissue - dorsal fillet. Remove liver	Freeze or analyse immediately	
M.edulis	Keep cool, and moist in a clean container	No. Time between collection and depuration must not exceed 24 hours	Scrape off growth on shells and scrub with nylon brush and rinse with clean water	24-48 hours in clean seawater. 48 hrs if mussels are from a turbid environment. Change water 2-3 times every 24 hrs. Remove byssal threads following depuration	Deep frozen	Remove shells, use flesh only	Freeze or analyse immediately	
O.edulis	Keep cool and moist in a clean container Collect seawater from the site for use in depuration	No. Time between collection and depuration must not exceed 24 hours	Scrape off growth on shells and scrub with nylon brush and rinse with clean water	48 hours in settled seawater for the site of collection, change water every 24hrs	Deep frozen	Remove shells, use flesh only	Can be frozen but best analysed immediately	

C.edule	Keep cool and moist in clean buckets with some sediment	Kept cool and in sediment (up to 24 hrs only)	scrub with nylon brush and rinse with clean water	3 to 4 days in clean seawater, change water every 24hrs	Deep frozen	Remove shells, use flesh only	Can be frozen but best analysed immediately	Bryan *et al* (1980)
P.vulgata	Keep cool in buckets with some water from the site	No. Process within 24 hrs	Scrape off growth on shells and scrub with nylon brush and rinse with clean water	N/A	Deep frozen	Remove shells, use flesh only	Can be frozen but best analysed immediately	Bryan *et al* (1980)
L.littorea	Keep cool in buckets with some water from the site and seaweed fronds if available	No. Process within 24 hrs	Scrape off growth on shells and scrub with nylon brush and rinse with clean water	N/A	Deep frozen – although not recommended	Remove shells, use flesh only	Can be frozen but best analysed immediately	Bryan *et al* (1980)
N.diversicolor	Keep cool and moist in clean buckets with some sediment	Kept cool and in sediment (up to 24 hrs only)	Gentle washing (in fine sieve)	6 days in acid-washed sand 1 day in clean seawater only. Changes sand and water every 24hrs	Deep frozen	N/A (Whole animals)	Deep frozen	Yorkshire Water Authority (1984)
Brown Shrimp (*Crangon crangon*)	Place shrimp in a clean polythene buckets with seal tight lids. Keep cool. DO NOT add seawater.	No. Process within 12 hrs	Wash in cold tap water to remove any debris	N/A	Deep freeze individually		Deep freeze in batches / samples	

On return to the laboratory cleaning of the specimens may usually be carried out with tap water, although clean seawater (if available in sufficient quantities) is better. Where cleaning is not followed by depuration, material should be shaken to remove surplus water, or blotted dry.

If depuration is required it should be carried out using seawater from the area of collection.

The effects of storage have not been well researched, and the recommendations in Table 4 are therefore only general guidelines on maximum periods. Long term storage leads to increasing risk of sample deterioration – freezers have been known to fail with the loss of all samples. Therefore, samples should be processed as soon after collection as possible.

Processed samples must be put in the pots provided by the National Laboratory Service.

5.1 Fucoid collection

Fucus vesiculosus (Bladder wrack) is the first choice. However, if it is unavailable, *F. serratus* is second alternative in open coast areas.

Collect by hand from rock surfaces just below the upper limit of the *Fucus* cover. Collect across an area to avoid denuding one area of all seaweed cover

Collect whole plants (from holdfast to apex of frond), break at the stipe or just above the holdfast. If populations cannot support this level of harvesting, fronds may be taken instead rather than whole plants. In very exposed areas or areas of low salinity, plants will be much smaller, in this instance, collect the largest available plants.

Do not collect detached or damaged plants or plants covered in epiphytic growth or ingrained with sediment.

5.2 Fucoid processing and preservation

Remove sediment and epiphytic flora and fauna by gently scrubbing with a nylon brush and rinsing with clean water. Ingrained material can be removed by running one blade of the scissors obliquely over surfaces of the frond.

Prior to dissection remove excess water from fronds by shaking them. Use stainless steel scissors or scalpel blade. Before dissecting fronds from different sites, replace scalpel blade or thoroughly clean scissors. Dissecting equipment should be kept exclusively for fucoids. Discard new tissue, thickened stipe, bladders and reproductive bodies, the remaining frond tissue is retained for analysis (see Figure 1).

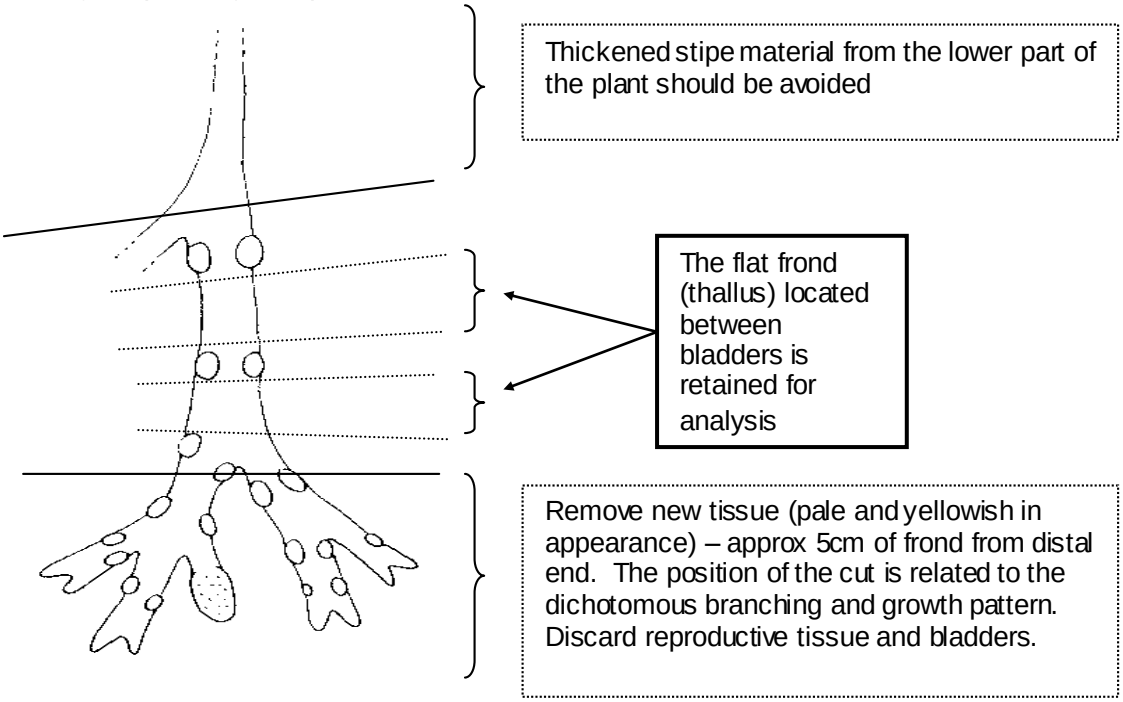

Figure 1. Dissection of fucoids

Cut frond tissue into approximately 5mm squares*, directly into a pre-tared sample container, record sample weight and seal immediately.

Equal quantities of material should be dissected from each plant, where possible 0.35-0.8g of frond tissue should be taken from each plant.

Processed material may be deep-frozen prior to analysis.

Record information on the Algal and Invertebrate Tissue Processing Data Sheet (Appendix 1).

Limited investigation indicates no significant difference between samples cut by scissors or samples which are blended in a clean blender. However, when

blending use short sharp blasts, as the heat created during blending may increase loss of volatile contaminants to the atmosphere.

5.3 Fish collection

Preferred target species are flounder (*Platichthys flesus*) and Dab (*Limanda limanda*). Plaice (*Pleuronectes platessa*) may be considered in areas where flounder and dab are absent. Whiting (*Merlanguis merlangus*) may also be considered.

Sampling should be performed at a time when impacts on the reproductive cycle of the target species will be at a minimum. As the breeding differs geographically workers should refer to OSPAR (1997) – JAMP Guidelines for Monitoring contaminants in Biota)[1], for spawning patterns of target species.

For collection methods refer to Technical Reference Material document - 011_07 Trawling for fish (otter, beam & oyster dredge).

When trawl comes on board the sample must be kept away from any possible source of contamination. All workers should wear clean gloves when handling fish, and samples must be deposited in clean containers.

Sort the catch as quickly as possible, return surplus and non-target species overboard (follow guidance on welfare of fish in Trawling for fish (otter, beam & oyster dredge). Make a record of fish which are visibly damaged or in a poor condition (the document 011_07 Trawling for fish (otter, beam & oyster dredge) provides a "Diseased Fish Record Sheet". Take care to differentiate between damage due to trawling and that caused by disease, damaged fish or fish in poor condition must not be selected.

There are two possible sampling strategies: sampling to minimise natural variability and length stratified sampling (see CSEMP (formerly NMMP) Green Book and JAMP Guidelines for Monitoring Contaminants in Biota). The sampling strategy selected must be standardised for a project, and employed consistently on subsequent sampling occasions. This means for specifically for CSEMP, sampling periods are defined as within 3 weeks either side of the original sampling occasion.

The size and number of fish required is dependant on the sampling strategy used. Suitable fieldsheets can be found in Appendix 2 in 011_07 trawling for fish technical guidance.

5.4 Fish processing and preservation

If dissection is to be performed at a later date it is recommended that whole fish are stored in a suitable container and frozen.

All dissections must be performed by trained personnel capable of identifying and removing the appropriate tissue.

Dissection should be performed using clean stainless steel knives (or scissors), which should be thoroughly cleaned between samples (if using scalpels, the blades should be replaced between samples). Clean disposable gloves should be worn whenever fish are handled, and these must be changed between samples, or hands must be washed.

[1] To access document go to "Measures" on the menu options and search for "contaminants in biota", select the appropriate reference to view document.

When dissecting muscle, in flat fish the eyed top fillet should be taken. In round fish, the right dorsal muscle should be removed from underneath the red muscle layer. The epidermis and any subcutaneous fat should be avoided. Dissected material should be stored in containers provided by NLS and frozen immediately.

Record all information on the Fish Tissue Processing Data Sheet (Appendix 2) then submit all key supporting data with the NLS submission sheet.

5.5 Shellfish collection

Collection of shellfish should be carried out at certain times of year to avoid variation in samples due to reproductive and growth factors. Table 3. gives a guide as to the most appropriate time of year. The month of collection should remain constant year on year for temporal studies.

Shellfish collected by hand should be taken from the mid-shore:

- Mussels and limpets - collect by hand using a stout blade (e.g. oyster knife) to prise them from the substrate
- Winkles - pick off by hand.
- Cockles - extract from sediment by hand after overturning sediment with a fork or spade (back fill holes afterwards).
- Oysters - collect by hand or catch using an oyster dredge, the oyster catch must be kept free of any contamination when bring dredges on board, all workers should wear clean gloves to sort through the catch quickly, returning any surplus overboard. Avoid harvesting dead individuals.

The shell length should conform to those in Table 3, however, those collected should all be within 5mm of each others length. For temporal studies, this narrow size range should be standardised and adhered to in subsequent sampling.

Sufficient number of specimens must be collected, as per Table 3. However, it is recommended that more are taken to compensate for any dead or under/over sized specimens inadvertently harvested.

The time between collection and depuration in the laboratory must not exceed 24 hours. See Table 4 for how samples should be transported to the laboratory. In the case of oysters, seawater from the site also needs to be collected and returned to the laboratory, settled and used during depuration.

5.6 Shellfish processing and preservation

Dead individuals must be discarded

Scrape off epifauna attached to shells and remove detritus and sediment by scrubbing with a nylon brush, rinse under cold tap water.

Mussels, Oysters and Cockles need to go through a process of depuration. The depuration process requires seawater; sufficient seawater should be collected from the site shellfish were collected from.

Place specimens on a rack raised off the bottom of a holding tank, and cover with clean seawater of appropriate salinity. Depurate for the recommended length of time (see Table 4). The system should be aerated and kept cool

throughout the depuration period. Change water as frequently as recommended in Table 4. Remove dead shellfish as often as possible.

Weigh (if required), and measure shellfish lengths (in the case of limpets measure across the greatest width. Discard under/over sized (re lengths) individuals. Samples may be frozen at this point, although they must be thawed prior to further processing (it should be noted that freeze/thawed mussels, cockles are easier to shell, however, winkles become difficult to work with).

Split into batches. Each batch should have a minimum number of individuals (to smooth out natural variability), follow Table 3. Sizes should be kept as close as possible between each sample.

- Mussels & Cockles - open by sliding an oyster knife blade between the valves and cutting the adductor muscle. Extreme care should be taken when dissecting, suitable protective gloves (preferably with kevlar) should be worn with disposable waterproof gloves. Let fluid inside the shell cavity drain away by placing the opened shell on absorbent paper for about 5 minutes.
- Limpets – remove tissue from the shell using an oyster knife, protective gloves (with kevlar) should be worn with disposable waterproof gloves.
- Winkles – remove tissue by cracking the shell and extracting the flesh with stainless steel forceps (a bench vice can be used for added control)

Remove the flesh and weigh individually (if required). Bulk together and weigh the flesh from each batch. If required, individual shell weights and/or the total weight of the shells (i.e. no flesh) from each batch can also be recorded.

Flesh can be frozen pending analysis. The type of container used for storage will be the same as that sent out by NLS. Containers must be free of any contamination.

All equipment should be thoroughly cleaned between samples.

All processing details should be recorded on the appropriate Algae and invertebrate Tissue Processing Data Sheet (Appendix 1), then submit all key supporting data with the NLS submission sheet.

5.7 Ragworm collection

Collection of ragworms should be done in accordance with the guidance given in Table 3.

To collect ragworms sediment will require digging using a fork or small spade, depending on its nature. Individual worms can be removed from the sediment by hand or with forceps.

Only individuals < 200mm in length should be selected, as larger worms may have been part of the spawning population and could influence the results of analysis due to their probable poor condition. The method of collection will in effect miss juveniles (< 20mm) and so avoid their presence in the sample.

100 suitable individuals are required per sample, it is recommended that 150 are collected to compensate for oversized individuals and any subsequent deaths during transport and processing.

Follow guidelines in table 4 for transport of samples back to the laboratory.

5.8 Ragworm processing and preservation

Wash samples gently in a 0.5mm mesh sieve. Extract ragworms with plastic forceps. Any oversized, damaged or dead ragworms should be discarded.

Ragworms should be counted and transferred to suitable containers such as trays, containing 2cm depth of acid washed sand covered with the same depth of filtered seawater diluted with de-ionised water to the same salinity as that at the collection site.

Cover trays and refrigerate at 4^{o}C. The sand and water should be changed every 24 hours, and any dead worms discarded. After the fifth change the worms should be transferred to clean water for a further 24hrs, after which they are counted, bulked into batches and weighed. Worms may be frozen pending analysis, although only refrigeration is required if analysing immediately.

Use containers provided by NLS. All processing details should be recorded on the Algae and invertebrate Tissue Processing Data Sheet (Appendix 1)

5.9 Brown shrimp collection

Shrimp may be collected by a variety of methods (Oyster dredge, Naturalist dredge, beam/otter trawl).

The catch must be kept free of any possible sources of contamination. Workers must wear clean gloves.

Sort the catch quickly, retaining shrimp in the size range and numbers detailed in Table 3. Pick shrimp out by hand and place in clean containers. Return surplus catch overboard.

Transport shrimp in clean polythene buckets with seal tight lids. DO NOT add seawater, DO keep cool.

Process or freeze within 12 hours of collection.

5.10 Brown shrimp processing and preservation

Wash in cold tap water to remove any debris

Sort by size, keep the size distribution within replicates as even as possible and measure each shrimp individually.

If there is a delay in analysis, shrimp replicates can either be homogenised, the frozen, or frozen as a complete batch/replicate. The type of container used for storage will be supplied by NLS. Containers must be free of any contamination.

All processing details should be recorded on the appropriate Algae and Invertebrate Tissue Processing Data Sheet (Appendix 1).

5.11 Essential data steps

When batching samples, it must be possible for a reporter to link the replicate or batch with the survey information and the sample sent to the laboratory. So all samples submitted to NLS must have a batch number or description that relates to the length class or other category required. Without this the chemical data and the field and key supporting data cannot be matched.

Key data from the laboratory or field must be entered on the NLS submission form. This includes, mean shell weight, mean length/weight of fish, sex (if appropriate), This avoid mismatching later, and speed up the reporting process by RSU teams.

If required data is not entered on the NLS submission sheet, then it must be made available to those reporting the data. This is particularly important for CSEMP, where the sampler and data reporter are rarely in the same team.

References

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- Butler, P.A., Andren, L., Bonde, G.J., Jernelov, A., & Reisch, D.J. (1971). Monitoring Organisms. **In:** (ed. M. Ruivo) FAO Technical Conference on Marine Pollution and its Effects on Living Resources and Fishing, Rome 1970. Supplement 1: Methods of detection, measurement and monitoring of pollutants in the marine environment. Fishing News (Books) Ltd., London. pp101-112.

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- Topping, G. (1983). Guidelines for the use of biological material in first order pollution assessment and trend monitoring. DAFS, Marine Laboratory, Scottish Fisheries Res. Rep., 28, 28pp.

Appendix 1

Algal and invertebrate tissue processing data sheet.

N.B. Complete in ink and initial and date any corrections.

Survey Name/code:	
Species:	
Date and time Sampled:	
Sampled by:	
Date Processed:	
Processed by:	

Site Name/ Code	Batch number	Length range in batch	Number of individuals in batch	Weight of batch(g)	Weight of Container & Sample (g)	Wet Weight of Sample (g)	Shell weight (g) (shellfish only)

General comments:

Appendix 2

Fish tissue processing data sheet.

N.B. Complete in ink and initial and date any corrections.

Survey:		Species:	
Site:		Date Processed:	
Date and time sampled:		Processed by:	

Fish Length (mm)	Sex	Tissue Type	Weight of Container (g)	Weight of Container & Sample (g)	Wet Weight of Sample (g)	Comments

General Comments:

Related documents

- 37_04 GRA - Field work
- 725_06 Fieldwork
- 600_06 Lifejackets and buoyancy aids
- 426_05 GRA - Working in or near water
- 732_06 Working in or near water
- 193_06 GRA - Intertidal Soft Sediment,
- 13_07 Intertidal Soft Sediment
- 32_04 GRA - Boatwork.
- 730_06 Working from a boat,
- 767_06 The Safe Management of Boatwork
- 242_06 GRA - Ecology Laboratory Safety,
- 52_05 Ecology Laboratory Safety
- eLearning - Understanding Tides and Tide Tables
- Notes on ground data requirements for CASI remote sensing surveys, provided by Science. Guidance on Position-fixing using Hand-Held GPS (in draft)
- Sampling Procedure (Environmental and Effluent) CD – Chapter on General Sampling Procedures
- Marine Ecology Monitoring CD – Chapter on Bioaccumulation
- Fisheries Monitoring CD – in preparation (see Training & Development site on easinet)
- eLearning - Understanding Tides and Tide Tables