Report

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SAMPLING MACRO-INVERTEBRATES ON INTERTIDAL FLATS TO DETERMINE THE POTENTIAL FOOD SUPPLY FOR WADERS

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Introduction

The main purpose of this document is to describe procedures developed over the last 30 years (McGrorty, 1973) by members of the behaviour-based modelling group (BBMG) at CEH Dorset, for sampling different components of the invertebrate macro-fauna of intertidal flats.

These consist of

1) A 'General Intertidal Survey Procedure' used to survey the abundance and distribution of all the main macro-invertebrate groups and/or species present on an intertidal flat.

2) A 'Cockle Survey Procedure' used to survey the abundance and distribution of cockles, or any other near-surface dwelling bivalve.

3) A 'Mussel Survey Procedure' used to survey the extent of, and mussel density and percentage cover within, mussel beds.

These procedures are used by the BBMG to supply the information needed to characterise the food supply in our individuals-based wader models, but as they provide information about the overall amount of biomass of different species present and its spatial distribution, they are of value in their own right.

Health and Safety

The responsible person for any team using the following guidelines to sample intertidal or wetland invertebrates should assess the risks of local circumstances and put in place procedures to minimise them. In European Union member states you should follow local regulations based on EU Health and Safety directives. Elsewhere, the current laws, regulations and procedures should be strictly followed to assess and minimise potential risks.

General Intertidal Survey Procedure

Survey design

This procedure is designed to produce estimates of the population size, spatial distribution and size-structure of important wader prey species within a site. Different wader species feed on different size ranges of different invertebrate prey species (see Table 1, adapted from Goss-Custard (In prep.)), so estimates of the size-structure and biomass density of these species are essential for parameterising CEH's individuals-based wader model[1]. Species-level identification of very small and/or rare organisms is not important for this purpose, so this procedure emphasizes the detailed measurement of important bird prey species rather than any need for precise, species-level identification in all cases.

[1] It should be noted that, although we consider this sampling scheme the most effective way to supply information for CEH's wader models, the models can also be developed using data collected by other sampling methods e.g. (Stillman, et al., 2003)

The survey has four main aims

1) To map the distribution of all the main wader prey species in a site

2) To determine the total population of each species or prey type

3) To determine the size structure of those populations

4) To determine the biomass of those populations

There are two ways in which these aims could be accomplished. One is to use an initial survey to map the distribution of each species/prey-type, then to use a stratified random sampling scheme to characterise the populations mapped in the initial survey. The other is to combine mapping and sampling in one survey by using a systematic grid-based sampling scheme.

The advantage of the two-stage mapping/sampling approach is that it allows for stratified random sampling of prey populations, which in turn allows statistically unbiased error estimation and thus more precise estimates of the mean population density of each species. There are several disadvantages of this approach. First, it requires two surveys so is more labour-intensive, particularly as the initial survey would require sampling to establish the presence/absence and density of many species that leave no indication of their presence at the surface. As intertidal invertebrate populations are often very variable in their numbers and distribution, it is unlikely that as single initial survey would provide an adequate basis for stratification in repeated subsequent surveys. Finally, it may be difficult to design a stratified sampling scheme suitable for a number of species whose distributions overlap to a greater or lesser extent.

The advantages of using a systematic grid-based approach are i) it can be carried out without pre-survey or extensive prior knowledge about faunal distribution[2] ii) it provides a good map of species distribution iii) it is much easier to carry out in practice and iv) as samples are spread evenly across the whole area, sampling sites do not have to be changed from survey to survey. The main disadvantages of grid-based survey are i) because all subsequent samples are fixed relative to the position of the first sample, estimates of mean density will be biased in the statistical sense, although in practice not likely to be further from the true population mean than a random sampling survey and ii) if there is zonation within the site which matches approximately the size of the grid, there is a danger of missing some species altogether. It is currently unknown how often the latter disadvantage is likely to occur on intertidal flats, but to some extent it can be overcome by using an asymmetric grid based on the most important perceived environmental gradients, e.g. sampling at smaller intervals downshore than alongshore. However the grid is arranged, it is important to remember that one of the main aims is to produce a map of species distribution over the whole site and to ensure the selected sampling scheme can provide this.

[2] This is not to say that knowledge of conditions on the site itself is not required. For safety reasons, as much local knowledge as possible should be sought before surveying and unknown location.

Normally, the practical starting point for planning the survey is to estimate how many samples can be taken within the time and budget available, taking into account the considerable time needed for processing each sample after the fieldwork is complete.

In our experience, the average processing rate is approximately 4 samples per day in the lab. Although the time taken for individual samples varies considerably around this figure, the average should remain more-or-less fixed given the protocol.

The rate at which samples can be taken in the field is very variable, depending on how accessible the sample locations are, the mode of transport used to reach them and the timing of low tide in relation to daylight hours at the particular site being surveyed. As the best time for surveying is limited to a few weeks in autumn (see next section) the fieldwork is best achieved by bringing together as many people as possible for a concentrated effort over a short period of time.

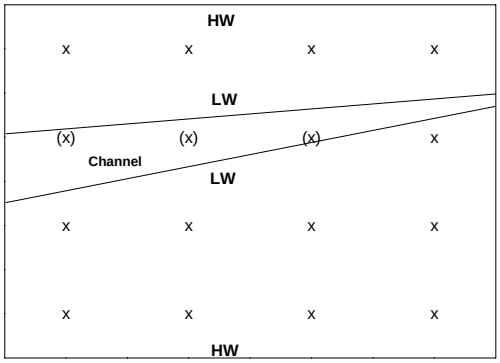
The size and positioning of the grid depends very much on the nature of the site being surveyed. CEH has used both symmetric and asymmetric grid sampling schemes successfully at a number of sites throughout the UK, ranging from a completely symmetrical 0.25km grid on the Exe estuary (Durell, et al., In press) to a highly asymmetric grid on the Wash (Yates, et al., 2002).

Generally, we base our grid on that on the Ordnance Survey maps. We mark a cross (representing a sampling point) at each 1km grid intersection that falls in the intertidal area, then at each 0.5km intersection (first downshore, then alongshore – See Fig. 1), then each 0.25km intersection and so on, until we reach approximately the number of samples we have estimated can be taken[3]. Grid intersections which are near, but not quite on, the flats are included because OS maps are rarely an up-to-date representation of the intertidal areas. This means that samples are restricted to those sites at or above mean low water. This is adequate for the purpose of model parameterisation as sites below this level are seldom available to waders. If lower levels are required the grid can be based on the extent of banks/flats shown by Admiralty yachting charts which extend down to the lowest astronomical tide.

In large sites, like the Wash, a symmetric grid would not be the best option as the large distances along shore mean little information about downshore variation would be captured. In these cases an asymmetric grid would be preferred. The size of the grid can be arrived at by deciding the appropriate distance between samples required to capture downshore variation, then setting the grid interval along-shore to an appropriate value based on the number of samples that can be taken. This is, in fact, transect sampling, but because distribution maps must be produced it is essential that the transects are evenly-spaced along the shore and *not* concentrated around access points.

[3] 'Filling in' of the grid should be done in blocks, e.g. fill in all the 500m intersections downshore, tally the number of samples and, if insufficient, fill in all the 500m intersections alongshore. Clearly it makes no sense to stop at exactly the required number of samples and leave some parts of the grid at a different resolution to others.

(a)



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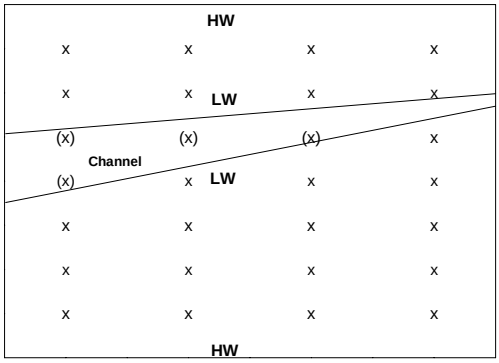

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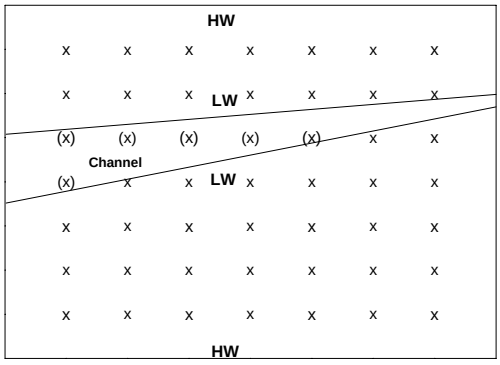

Figure 1. Filling in from a 1km grid. The rectangles represent an estuary with HW and LW representing high and low water marks respectively and the river channel shown by diverging lines just above the middle. Crosses are grid intersections. Those that fall in the channel (in parentheses) are unlikely to be sampling points, but surveyors on the ground should check to ensure the channel has not moved and made these sites available. a) 1km grid. b) Filled in downshore to make a 1km by 500m grid. c) Filled in alongshore to make a 500m grid.

Survey Timing

The survey should be conducted in September, extending into October if necessary. This is the time of year when migratory waders have just returned to their wintering grounds and when prey biomass is at its post-summer maximum, prior to the onset of senescence and depletion. In western Europe, large spring tides also occur in daylight during these months, allowing the best access to the birds' intertidal feeding areas.

Sampling Procedure

Sampling points should be located using a GPS or map, pacing stick & compass. On arrival at the sampling point care should be taken should be taken to avoid conscious or unconscious bias when placing the corer. For example, the sample could be taken at the tip of the toe after the last pace if walking to the site, or 1m in front of the vehicle if travelling by quad-bike.

If you can get close, but not actually to, the exact sampling point, a sample should be taken as close as possible to the correct point and it's position recorded for future reference.

At each sampling point:

1. to sample very small and/or abundant species, a 10cm diameter sediment core is removed to a depth of 30cm. The bottom half of the core is broken up and sorted by hand to locate the large invertebrates that might occur at this depth. If transport is available these are placed with the top half of the core in a labelled plastic bag for further processing later. If on foot, partially sieve the top half of the core by placing in a large polythene bag with sea water, shake gently to liquidise and pour through a 0.5mm mesh. Repeat 2 or 3 times if necessary, then wash the contents to the corner of the sieve and transfer to the labelled bag. This greatly saves on the weight of wet sediment to be transported without any loss of data

2. To sample large burrowing worms such as Arenicola marina, a randomly placed 1x 1m area is marked out on the surface and the numbers of worm casts, tubes or holes within it counted. If there is any doubt about the identity of these a nearby area should be dug up to check the species[4].

3. To sample large, less abundant species which are unlikely to occur at high enough densities to be sampled effectively by the coring technique, one quarter of the $1m^2$ area is marked off and any larger surface-dwelling molluscs present (e.g. mussels and winkles) within that area counted and collected. This area is then hand-raked or, if water is available nearby, dredge-netted with a 2mm mesh net to count and collect other near-surface dwelling molluscs (e.g. cockles). These samples should be frozen on return to the laboratory.

4. Records are made of any noteworthy features e.g. surface sediment features, depth of aerobic layer, species of plant or alga present, e.g. Zostera,

[4] If there are significant numbers of such worms present in the study area as a whole, some worms should be dug up from a selection of sites and measured to determine the size distribution present.

Enteromorpha, Ulva, Fucus and their percent cover if possible in the quadrat and in the general vicinity. Vegetation within the quadrat may help to explain otherwise inexplicable low values of invertebrate abundance (e.g. anoxic conditions under algal mats) and data on the wider presence of vegetation around each sampling station may provide valuable information on the abundance of food supplies of herbivorous wildfowl.

5. If specimens are being collected at the same time for ash-free dry mass (AFDM) estimation, a few (2-3) of each of the main species present at the sampling point should be placed individually (one specimen per bag) in small Ziploc plastic bags. These should be frozen on return to the laboratory or within 24h of collection, whichever is sooner. Note, worms should be measured before freezing.

As soon as possible after collection, each sample should be sieved through a 0.5mm nylon mesh sieve, nylon being less destructive than brass (see Appendix 1 for the sieve design we use). The contents should then be fixed in a solution of 4% formalin (40% *buffered* formalin diluted with 1:9 seawater). If the samples cannot be sieved on the day of collection, they should be stored at 4ºC and processed within 24 hours.

Once fixed the samples should be washed in fresh water and preserved in industrial methylated spirits (IMS) prior to processing.

Processing

The sieved and preserved samples are washed again in the laboratory through a 0.5mm nylon mesh to remove residual sediment.

All macro-invertebrates in each sample are counted and identified to the level of taxonomic detail that is necessary to quantify the abundance of the various important types of bird food. This is generally species level for all except ‘small worms’, e.g. oligochaetes, capitellids, spionids etc.

Some worms are likely to be broken either during the coring or sieving process. Only the head end of broken worms should be counted.

The maximum length of all individuals of all species that are above the minimum size taken by any of the bird species of interest are also be measured to the nearest mm. There are two exceptions to this: 1) intrinsically small species, e.g. ‘small worms’, which it would be too time-consuming to measure and in any case unnecessary for modelling purposes and 2) mussels, which are measured using length parallel to the ventral surface (Fig. 2), usually, but not always, equivalent to maximum length.

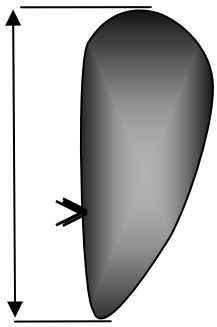

Figure 2. Measuring mussel length

The length of any broken worms should be estimated from jaw length, if present, or roughly from the breadth of the remaining front part in comparison with intact specimens present. If this is not possible, the worm should be recorded as 'broken, length unknown'

If a sample contains excessive numbers of any species, only a sub-sample should be counted. The method of sub-sampling is best judged on a sample-by-sample basis, but details of any sub-sampling methods used should be provided with the data.

Ash-free dry mass estimation

For each of the main prey species an ash-free dry mass (AFDM) length relationship is required to calculate the biomass density at each sampling site[5]. It is also needed to calculate the potential intake rate of the birds in the model as this depends on the mean AFDM of the prey they are eating.

The individual animals used to derive these relationships can be collected during the survey, but should not be extracted from the main set of samples as this can easily lead to confusion. For each species, c. 50 individuals that span the entire range of sizes present should be collected. In the case of molluscs and crustaceans the 50 individuals can be placed together in one bag and the frozen prior to further processing. Worms, however, should be measured while alive and then frozen individually, as they thaw rapidly during processing and leak body fluids. The length of the worm should be written in waterproof marker on the bag in which it is frozen. Worm lengths should be measured when the worms are 'relaxed' being neither concertinaed up nor stretched out. There is an element of subjectivity in this but, in our experience this still produces good AFDM-length relationships with a high R^2 value[6].

[5] Although it is preferable to collect AFDM relationships at the time of the survey, the model can be parameterised using values from previous estimations or published estimates from similar sites.

[6] In ragworms, the jaw-length can be measured after freezing and thawing. Although this is, on the face of it, less subjective than measuring body length, in fact measurement error and variation in the

To determine the AFDM of each individual, first measure the length as follows:

- Mussels – parallel to the ventral surface, as in Fig. 2
- Other bivalves – maximum length
- Worms (excluding 'small worms') – 'relaxed' body length (see above)
- Crustaceans – body length from tip of rostrum to telson
- Gastropods – spire height

Lengths should be measured while the specimens are still frozen, using suitable gloves or implements to avoid chilblains.

Worms and crustaceans can then be placed into individual crucibles. Any body fluid or flesh remaining in the bag in which a worm was frozen should be washed into the crucible using water and a fine-jet wash-bottle. Small threadlike worms are very difficult to measure accurately and weigh too little to be ashed individually. In this case 50 individuals should be counted into a single crucible, ashed as described below, and the AFDM divided by 50 to calculate the AFDM of an average small worm.

Bivalves and gastropods (with the exception of Hydrobia, where this is impractical) should be placed in individual crucibles and allowed to thaw partially. Then, a suitable implement should be used to remove all the flesh from the shell, holding the item over the crucible to retain any body fluids that leak from the shell upon opening. Again, any remaining body fluid or flesh should be washed from the shell into the crucible using water and a fine-jet wash-bottle. The shell can be discarded unless required for other measurements.

The number or identifying mark of the crucible, length of the item and, if required, its age, should be noted in a table with additional columns for the weights.

The crucibles plus flesh should be placed in a suitable holder/tray and dried at 90^{o}C to constant weight. Usually 24 hours will suffice, but large 'fat' individuals might need longer. After the initial period of drying, remove the tray from the oven using heat-proof gloves and as quickly as possible transfer the crucibles to a desiccator using large forceps or tongs. Ensure that the silica crystals in the desiccator are blue rather then pink prior to transferring the specimens to cool. Note that the dried flesh (and probably the crucible) is hygroscopic and will quickly absorb moisture and gain weight if exposed to the air.

When cooled to room temperature, weigh the crucibles on a balance accurate to 0.1mg, note the weight (crucible + dry flesh mg) and return to the desiccator. Return to the oven for a further 3-4 hours and repeat the procedure until there is no further weight loss (i.e. the only difference is in the fourth value after the decimal point). The

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curvature and degree of wear of the jaws mean that the AFDM/jaw-length relationships are no more accurate than those obtained by measuring body length.

accumulated drying time can be used as a standard for other sets of similar sized samples.

Using gloves and long tongs transfer the crucibles to a muffle furnace and burn at 550^{o}C to constant weight, normally for 4-6 hours. Remove the crucibles and place in a desiccator, allow to cool to room temperature and weigh. Note the weight (crucible + ash mg). Repeat the procedure, burning for periods of 2 hours until there is no further loss of weight. Note that the ash may also be hygroscopic so the use of a desiccator at all stages is very important. It is also important to 'refresh' the silica gel in the desiccator regularly; any sign of pink and the sample might absorb moisture from the crystals.

Note that by drying the crucibles first thoroughly in a muffle furnace at 550^{o}C and weighing empty, then whole fresh weight, flesh weight, dry flesh weight and ash-free dry weight can all be obtained in sequence by subtracting the weight of the crucible from the weight of crucible plus cockle at each stage in turn.

Subtract the crucible + ash weight from the crucible + dry flesh weight for the ash-free dry mass (AFDM) in mg. Regress AFDM (mg) against length (mm) transforming values to natural logarithms (ln or $\log_e$) and adding a quadratic term, if significant, to explain the maximum variance in weight due to length i.e. to maximise R^2. Note that an error is introduced by back-transforming predicted $\log_e$ AFDM values. To counter this, half the regression error mean square (EMS) must be added to the logged AFDM value before it is transformed back to a predicted AFDM in mg.

Sample archiving

If archiving is required, all of the contents of each sample processed for the purposes of identification and measuring (including detritus) are stored together in a single pot containing stabilised industrial methylated spirits (IMS), and labelled internally and externally with site and date. These pots are stored in sealed plastic boxes. Separate storage of individual species and of juveniles for each sample requires the unnecessary use of considerably more resources (plastic pots, IMS, storage space).

Sampling procedure for a survey of only cockles or other near-surface dwelling bivalves

Introduction

Fisheries scientists routinely survey cockles to determine stock levels and allowable fishing quotas. These surveys are easily adapted to provide the data for CEH's wader models.

The most important difference between a survey designed purely for fisheries purposes and one suitable for wader modelling is that the former concentrates only on known cockle beds, whereas the latter must encompass the whole of the intertidal area, as patches of cockles which are unprofitable to fish but nonetheless useful to birds often occur outside the main beds.

The sampling procedure described here would replace that described under 'General Intertidal Survey Procedure' above when only cockles and/or other surface-dwelling bivalves are to be sampled.

Sampling procedure

Following the ebbing tide locate the first site using map and compass and /or GPS[7]. If on foot, on arrival at the grid intersection / sample site place the $0.1m^2$ sieve squarely at the tip of the toe after the last pace to mark out an area of $0.1m^2$. This is to avoid any worker bias; consciously or unconsciously the quadrat may always be placed where there are more or less cockles (if they are 'squirting' or 'winking'), or where the sediment is wet rather than dry, smooth rather than rippled etc., which might affect cockle density. If using a vehicle, stop as close as possible to the grid references then take one pace beyond the front of the vehicle and place the sieve as before.

If the exact sampling site cannot be reached, for example for safety reasons, a sample should be taken as close as possible to the correct site and its position recorded for future reference[8].

Using the tool of your choice (hand-rake, trowel, spade) remove the top 5cm of sediment from the area marked out by the sieve and place it into the 2mm sieve. Sieve the sample in water either in the sample hole or a nearby pool or creek. Then place the cockles in a polythene bag labelled with the site number and the date.

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[7] Using the OS grid means that after the first sample all subsequent sites will be either north, south, east or west. Even using a GPS it can still help and save time if, using a hand-held sighting compass, landmarks are identified on the horizon in each direction as a guide.

[8] Below the high water mark maps/charts are rarely up to date. Even using boats/vehicles, because sandbanks and channels move about and salt-marsh advances and retreats, some sites may be inaccessible, while others may have become accessible. Either be prepared to sample any site which is unexpectedly exposed or note its presence for the next survey. In either case it is always advisable to carry spare polythene bags (+10%) as it is easy for one to be blown away or ripped by a broken shell.

Place the sample bags in a large labelled polythene bag and rucksack if on foot or in a cool box if using a vehicle, if the weather is warm, or if the samples will be transported a long distance before being processed.

Record that you have sampled the site (or, if for some reason you are unable to sample a site record the site number and the reason it could not be sampled). Consulting the map and grid references walk/drive to the next site and repeat the sampling procedure.

Where possible, sieves, trowels etc. should be washed in fresh water at the end of the day to delay the onset of rusting.

Storing and processing of samples.

Cockles will not survive much more than 2 days in the back of a car even in a cool box. Treat the samples gently; cockles that are thrown to the ground, or shaken violently in a vehicle, tend to gape and may die sooner. So, either process the samples at high water / in the evening, return them to the laboratory every 1-2 days for processing, or arrange for the use of a freezer close to the field site.

Ideally the length of every cockle should be measured. If this is too time consuming, then sub-sample by measuring all of the cockles from every second, third or fourth bag (i.e. from sites 1, 3, 5 … or 1, 4, 7 …etc); **do not** choose bags with the fewest or the most cockles as this could bias the size (and age) distribution. Measure the length of each cockle along its longest axis using vernier callipers. Close the jaws gently to just touch the cockle, then rotate it slightly to find the maximum length. Read the value (mm) and record it, either on paper or directly into a spread sheet.

Approximately 60 cockles from the samples should be retained to determine cockle weight. If possible, retain three cockles (1 small, 1 medium & 1 large) from every other sample site. This ensures that all areas of the estuary and both high and low-density areas are represented. If there are, for example, no small cockles at a particular site, then only a medium and large specimen will be collected. Place the cockles from each site in a polythene bag and keep cool. At the end of each day's processing, sub-sample the retained cockles by taking a number of small medium and large cockles from the bag and placing each one in an individual Ziploc plastic bag. Freeze as soon as possible (before they begin to gape and die). The number taken each day should be proportionate to the number of samples processed that day, e.g. if the survey contains 400 samples and 100 have been processed in a day, 60*100/400 = 15 cockles should be retained and frozen from that day's samples.

If the age of the cockle is required, count the rings on the shell and record this also. Check that the number of rings is the same on both valves, as damage can result in a clear line on one but not the other.

Sampling procedure for a mussel survey

Introduction

In contrast with most other intertidal invertebrates, mussels are visible at the sediment surface, so a mapping and stratified sampling technique can be used easily and fairly rapidly in a mussel survey.

This field procedure is based on the methods developed by CEH for surveying intertidal mussel beds. It is designed to produce a map of the bed, a value of the percentage cover of mussels on the bed and an estimate of the mussel density and size-distribution. Full details can be found in McGrorty et al.(1990).

If a bed has clearly different, and discrete, areas of mussel density, high and low, these can be treated as two strata for mapping and sampling purposes. This should improve sampling efficiency and reduce variance.

Sampling procedure – Mapping

First, establish a baseline alongside the mussel bed using bamboo canes or find existing markers using a GPS or compass bearing.

If establishing a baseline for the first time, the position will depend on the shape and position of the mussel bed. Generally, the baseline should be established parallel to the longest axis of the bed.

If there are no existing marker posts, insert a crossed pair of bamboo canes at one end of the baseline. Record your position using a GPS. Establish the direction of the baseline by looking along it and taking a bearing on a distant object using a sighting compass. Walk along this bearing inserting bamboo canes at regular intervals (250m or less) until you reach the other end. Record the position of this end using the GPS.

The distance across the mussel bed is measured along a series of regularly spaced transects at right-angles to the baseline, using a 1m pacing stick. To obtain the inter-transect interval, divide the length of the baseline by the number of transects you intend to take *plus one*. The first transect can either be located randomly along the baseline within the first interval or at half the interval width. Starting at this transect, work out the bearing of your transects by adding or subtracting 90^{o} to/from the baseline bearing. Identify a distant object on this transect bearing and walk towards it using a 1m pacing stick. The distances at which the boundaries of the bed were crossed is noted. At each 1m pace across the bed the presence or absence of one or more mussels (>20mm) within a 20x20cm quadrat is noted. These data are used to draw an outline map of the mussel bed, to calculate its area and the percentage cover of mussels within it and hence the actual area of mussels.

When you reach the top of the transect, i.e. the end of the mussel bed, record your position, either in terms of paces of by GPS. Turn parallel to the baseline and walk [X]m to the start of the next transect then repeat the process until you have covered the entire mussel bed. (Fig. 3)

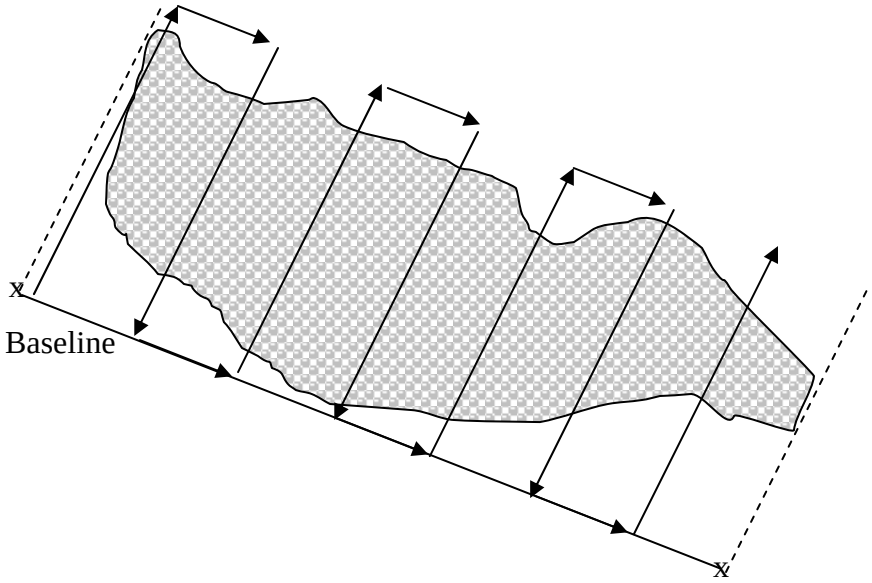

Figure 3. Mapping a mussel bed

Sampling procedure – Estimating density and % cover

To estimate mussel density, a number of samples (20x20cm quadrat) of mussels are taken at random from each bed.

The number taken is determined on site according to the area and the variability of the mussel cover / density within it. Samples are apportioned to mussel beds based on the total number that can be processed and the relative area of the beds.

At each sampling site, place a 20x20cm quadrat at the tip of your toe to avoid subconscious bias in quadrat placement.

Remove all the mussels within the quadrat. Place them in a plastic bag with a waterproof label stating the sample number, the mussel bed and the date on which the sample was taken.

If there are no mussels present, still place a label within an empty bag to record the fact that the sample was taken but contained no mussels.

If the mussels are very dense it may be necessary to remove mussels from only half of the quadrat. If you do this, make sure this is recorded on the label and preferably in a field notebook as well.

A separate sample of 40-50 mussels covering a wide range of sizes (20-70 mm) is also taken at random from each of the main areas to determine the ash-free dry mass of the mussels. Ideally, these should be taken a few at a time from a range of sampling sites to cover any environmental gradients across the bed.

Field procedure – rapid mussel survey

This procedure combines the mapping / %cover and sampling aspects of the above procedure in one set of transects. It is useful for giving a relatively rapid estimate of the extent and density of mussel beds about which there is no prior knowledge.

First, establish a baseline alongside the mussel bed using bamboo canes or existing marker posts. If a baseline has already been established in a previous year, use marker posts or GPS to relocate it.

If establishing a baseline for the first time, the position will depend on the shape and position of the mussel bed. Generally, the baseline should be established parallel to the longest axis of the bed.

Mark the start of the baseline with a bamboo cane and take a bearing along the baseline to a distant object.

Turn 90^{o} so you are perpendicular to the baseline and identify a distant object on the correct bearing. Walk along the transect towards this object, measuring progress with a 1m pacing stick or a GPS.

At regular intervals stop and take a $0.1m^2$ quadrat sample of the mussels, placing the quadrat at the tip of your toe to avoid bias. Arrange this so that, in effect, samples are taken on a grid across the whole bed.

Remove all the mussels within the quadrat. Place them in a plastic bag with a waterproof label stating the sample number, the mussel bed and the date on which the sample was taken.

If there are no mussels present, still place a label within an empty bag to record the fact that the sample was taken but contained no mussels.

If the mussels are very dense it may be necessary to remove mussels from only half of the quadrat. If you do this, make sure this is recorded on the label and preferably in a field notebook as well.

When you reach the top of the transect, i.e. the end of the mussel bed, record your position, either in terms of paces or by GPS. Turn parallel to the baseline and walk [X]m to the start of the next transect then repeat the process until you have covered the entire mussel bed (Fig. 2)

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Table 1 The size range of prey from which common shorebirds obtain most of their consumption. Values are minimum and maximum using CEH 1mm size classes; *i.e.* 1 - 29 means 1.000 - 29.999mm in terms of actual length. 'max' means the birds take sizes up to the maximum length present in the sediment.

	Mytilus	***Mya***	***Cerastoderma***	***Scrobicularia***	***Macoma***	***Hydrobia***	***Corophium***	***Hediste****	***Arenicola***	***Carcinus***	***Crangon***
Bar-tailed godwit	-	-	-	8-19	8-19	-	-	25-max	25-max	-	-
Black-tailed godwit	-	-	-	8-19	8-19	-	-	25-max	-	-	4-max
Curlew	-	25-max	8-19	20-49	8-max	-	-	50-max	50-max	10-39	-
Curlew-sandpiper /Dunlin	-	-	-	3-6	3-6	1-4	3-max	10-59	-	-	-
Grey plover	-	-	-	8-19	8-19	1-4	-	20-max	20-max	-	-
Knot	5-24	8-16	5-14	8-16	8-16	1-4	-	10-59	-	-	-
Oystercatcher	30-59	16-39	15-max	20-max	12-max	-	-	50-99.9	50-max	10-50	-
Redshank	-	7-13	-	7-13	7-13	1-4	4-max	15-79	-	3-7	4-max
Ringed/Kentish plover	-	-	-	-	-	1-4	3-max	10-49	-	-	-

* = and other worms too, such as *Lanice*, Cirratulids *etc*.

Appendix 1 Design for a 'McGrorty' floating sieve.

The sieves used for our surveys are constructed from wood and nylon (general intertidal survey) or heavy-duty stainless steel (near-surface dwelling bivalve survey) mesh. The advantage of this is that the sieves are relatively light and will not sink if accidentally dropped into water.

Materials (per sieve)

- Wood – planed softwood 4 each of 338x100x22mm and 338x22x22mm
- Mesh – Nylon 0.5mm aperture size or stainless steel, plain weave, 1.98mm aperture size, mesh count 10, 0.56 mm wire. Piece approx. 350x350mm
- Tacks – 12 and/or Staple gun
- Stainless steel screws – 20
- Wood glue
- Silicone sealant

Construction

- Glue and screw together the four side pieces, as shown in Fig. 1 to produce a square frame with internal dimensions of 316mm (area of $0.1m^2$). If the timber is not 22mm thick the lengths of the side pieces will have to be adjusted accordingly. Allow the glue to dry before proceeding further
- Metal mesh
    - Lay the mesh over the base of the frame and attach with three tacks per side (as in Fig. 2).
    - To avoid hazards from sharp protruding wires, fold over the edges of the mesh so they are tucked under the sieve. Use protective gloves or pliers for this stage.
- Nylon mesh
    - First soak the nylon mesh in water, or it will stretch and sag in use
    - Lay the mesh over the base of the frame and, starting on one side, staple in place every 2-3cm. Try to pull the mesh as taut as possible without causing distortion of the weave while attaching the staples.
    - When one side is stapled repeat the process on the opposite side, again pulling the mesh as taut as possible, then on the remaining two sides
    - Fold over the edges of the mesh so they are tucked under the sieve and secure with 2-3 more staples on each side.
- Glue and screw the 22x22mm battens to the base of the sieve to hold the mesh in place.
  Use three screws per side, ensuring that they are offset from the tacks or staples used to attach the mesh to the frame (Fig. 3).
  Also note that the joints of the side and base pieces are offset to increase strength (Fig. 1)
- Finally, seal all joints with silicone sealant, paying particular attention to the internal joins so none of the sample can escape around the base of the sieve.

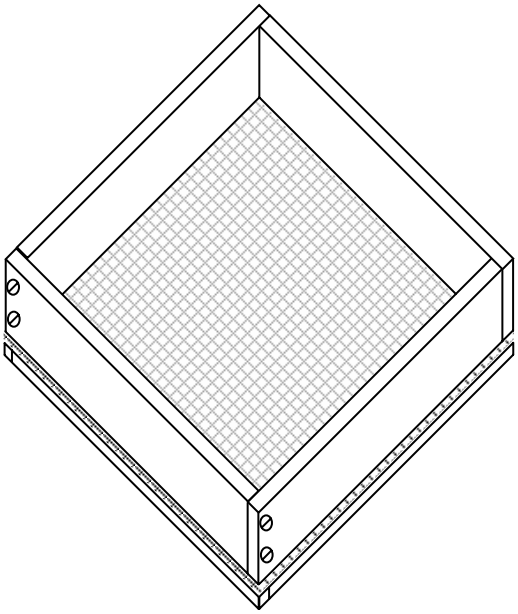

Figure 1. Finished sieve

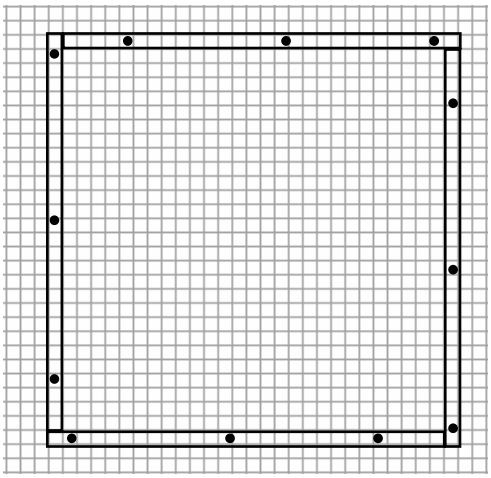

Figure 2. Attaching the mesh to the base

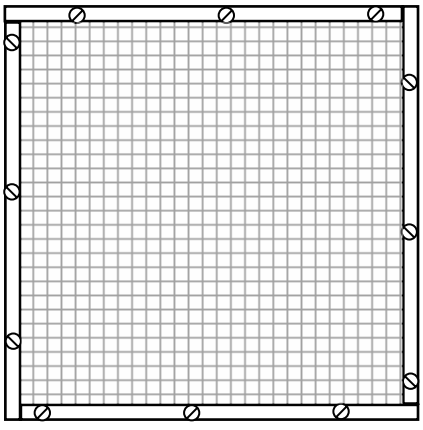

Figure 3. Attaching the base battens. Note offset of the screws from the tacks shown in Fig. 2 and offset of batten joints from main sieve joints.