

# Sampling soil for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; eraser; permanent marker pens; tape measure; 25 cm ruler; penknife; clean glass jars (500 ml capacity) with aluminium foil-lined screw-caps or strong polyethylene bags 30 x 20 cm (or similar); plastic-coated wire ties for securing the bags; clean water; detergent; acetone; paper towels; cloth; spade or soil auger; digging trowel; site map; compass; GPS (optional); cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass vessels being damaged or broken during transit; protective clothing; nitrile or rubber gloves.

Thoroughly check all equipment before you go into the field.

Identify the sampling site and mark on the site map for future reference.

Decide on the sampling approach based on consideration of the sampling objectives.

Wear protective clothing when sampling in pesticide-treated areas.

## SAMPLING WITH A SOIL AUGER/CORER

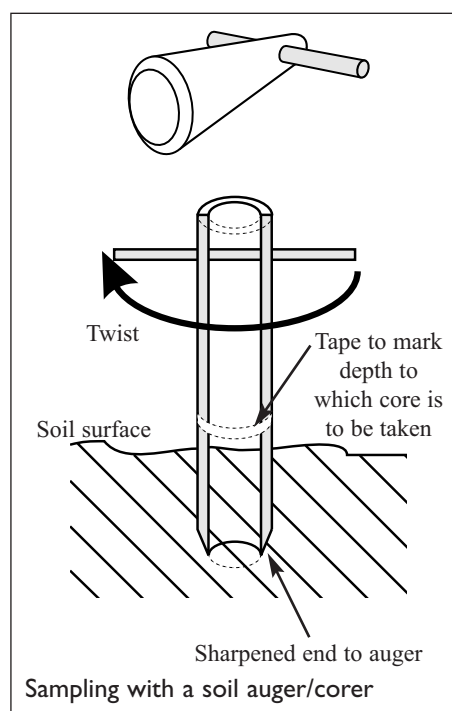
### Method

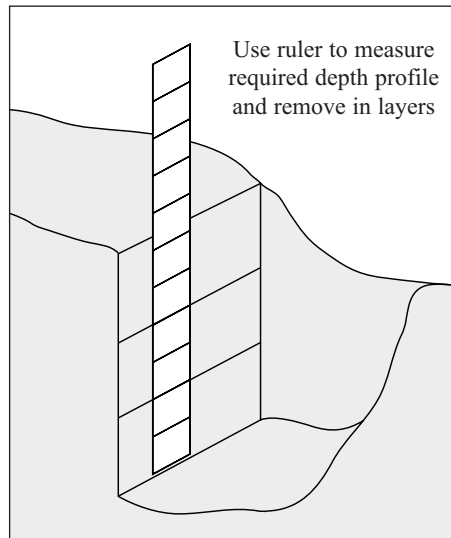
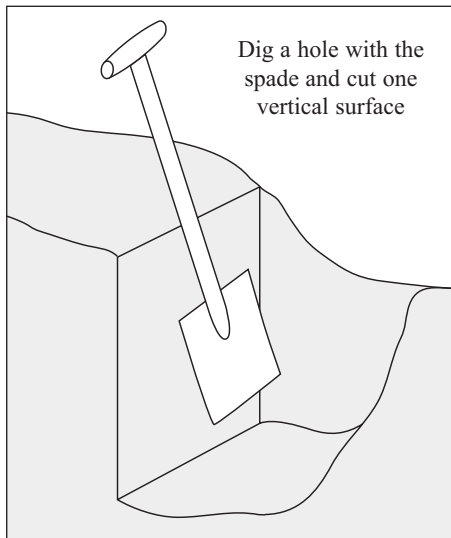
Collect samples by:

- composite sampling method: collect five cores to a uniform depth and mix together
- depth profile sampling method: take cores to a uniform depth, remove, extrude and section with a knife, taking 10-cm depth profiles. Take three replicate cores from each site, section and combine appropriate sections. The minimum sub-sample taken in this way should be 200 g (500 ml jar approximately half-full).

## SAMPLING SOIL FROM A DEPTH PROFILE

- If a soil auger/corer is not available, dig a hole to 30–50 cm depth with one side of the hole being cut vertically with a spade.
- With a ruler measure the required depth profiles and carefully remove the required layers (with spade or digging trowel), starting from the top (surface) layer (see diagrams over page).
- Again take replicates from each site and combine; the minimum sample size should be 200 g.
- Transfer the sample(s) prepared as above into a glass jar or wrap in aluminium foil and place in a polythene bag.
- Prepare a label giving the sample and site details and the date and put the label into the jar/bag. If using a bag, seal with a wire tie; if a jar, screw on cap.
- Place the sample jar or bag inside a second bag, prepare another label with all the relevant sample details. Put the label inside the outer bag and seal.
- Record the sampling details on the prepared data sheet (see page 143).
- Put the sample container into a suitable sample box for transportation. Protect with packing material.
- Clean the auger/corer and knife with water and detergent and then acetone before taking the next sample.





### OTHER CONSIDERATIONS

Use a cool-box if available to transport the samples. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where possible keep the samples chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

Always sample unsprayed areas first.

Change gloves between sample sites to avoid cross-contamination. Seal used gloves in a labelled plastic bag, until proper disposal can be organized.

# Sampling water for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string (various lengths including lengths up to 4 m); eraser; scissors; permanent marker pens; supply of clean glass bottles (1000 ml capacity) with teflon or aluminium foil-lined screw-caps; appropriate sampling device; dependent on depth of water at which sample is to be taken; long wooden pole; weighted cage to contain the sample vessel; wellington (rubber) boots or thigh waders (assuming sampling from the shore); rubber or nitrile gloves, preferably elbow length; cool-box where available or a strong sample box to contain the sample vessels after water collection and with appropriate packing (cardboard or foam rubber) to prevent vessels being damaged or broken during transit; protective clothing.

Thoroughly check all equipment before you go into the field.

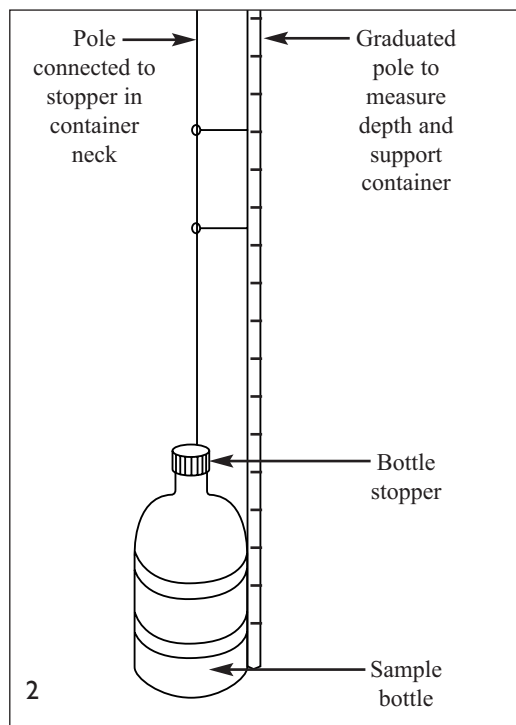
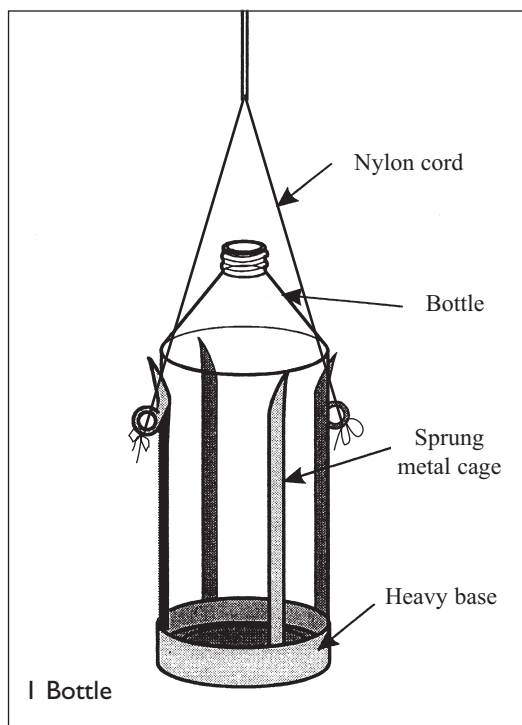
Select the sampling site and mark the site plan or map for future reference.

When sampling from close to the shore, and particularly where it is necessary to enter the water to find a sufficient depth in which to immerse the sample vessel, it will be necessary to ensure minimum disturbance to the bed of the river/stream/lake otherwise disproportionate amounts of sediment may be included in the sample.

The depth at which the water sample is to be collected should be determined in advance.

For surface or sub-surface water, the apparatus illustrated in diagram 1 should be used; for water samples at depths between 30 cm and 2 m (approximately) the apparatus shown in diagram 2 should be used.

Wear protective clothing when sampling in pesticide-treated areas.



## SURFACE/SUB-SURFACE WATER

### Method

- The depth of the water body will determine the technique used.
- In very shallow water the container (glass bottle) should be held in a nitrile gloved hand with the opening just below the water surface to allow it to fill.

- As soon as the bottle is withdrawn from the water it should be sealed with a clean screw-cap and an appropriate label attached such that all sample details are clearly presented.
- In slightly deeper water, the bottle, contained in a weighted metal cage, can be lowered by rope into the water (diagram 1). This is a useful technique when collecting a sample from a bridge or a boat. As soon as the bottle is withdrawn from the water it should be sealed with a clean screw-cap.

## SAMPLING WATER FROM A DEFINED DEPTH

### Method

- The apparatus in diagram 2 should be used.
- Lower the sampling device into the water to the required depth and remove the stopper using the central pole. Allow the container to fill.
- The central pole may be used to push the stopper back into place prior to withdrawal of the bottle from the water.
- Seal the container with a screw-cap fitted securely and attach an appropriate label such that all sample details are clearly presented.

### For all methods

- Record the sampling details on the prepared data sheet (see page 143) and give the sample a code number. Add that number to the sample label and, additionally, write the code number on the outer surface of the container with a permanent marker pen.
- Place the sample container into the sample box and secure it with packing material; ensure that it will not move or rattle against other containers during transportation.
- Clean the sampling device using water and detergent, rinse thoroughly and finally rinse with acetone. The outside surface of the bottles should be washed with clean water, dried and then labelled with the appropriate sample details. The metal cage or the pole mechanism should be washed with clean water or wiped with a cloth soaked in acetone, to remove any significant contamination which could be transferred when collecting the next sample. The apparatus need not be rinsed between the collection of replicates from the same site, only between sites.
- Repeat the sampling process such that a minimum of two replicate samples are collected.

## OTHER CONSIDERATIONS

Sample in unsprayed areas first.

If entering the water to take a sample, use the pole to check the depth and that it is safe to proceed. Watch out for the presence of crocodiles and beware of bilharzia. Ensure that adequate protective clothing is worn when working in water.

If, when entering the water to take the sample, sediment has been disturbed, it is important to allow this to settle before taking the water sample.

When using the sub-surface sampling equipment in diagram 1, the bottle will begin filling with water as soon as it is immersed and the sample will be a composite from water at the surface/sub-surface. When using the apparatus in diagram 2, the stopper is a rubber or cork bung which fits inside the opening of the screw-cap bottle neck.

The wooden pole or weighted cage can also be used to take samples from the shore where the water is too deep to allow wading or where the bottom sediment is soft (and dangerous) or is easily disturbed. Where possible keep the samples chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

Where a cool-box is available, this should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where sampling from a bridge or similar may be most convenient, the sample vessel should be tied to the wooden pole and lowered into the water or the vessel put into the weighted cage which is similarly lowered into the water.

Change gloves between sample sites to avoid cross-contamination. Seal used gloves in a labelled plastic bag, until proper disposal can be organized.

# Sampling sediment for residues

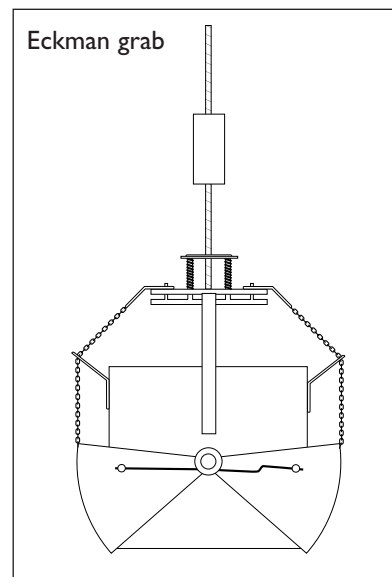
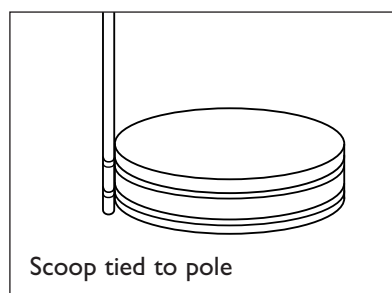
## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors; permanent marker pens; clean glass jars (500 ml capacity) with aluminium foil-lined screw-caps or strong polyethylene bags 30 x 20 cm (or similar); plastic-coated wire ties for securing the bags; sampling tool (scoop on a pole, core-sampling device or similar); folding ruler; wellington boots or waders; rubber or nitrile gloves; 2 m wooden pole; site map; compass; tape measure; clean water; detergent; acetone; cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass vessels being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Identify the sampling site and mark the site map.  
Wear protective clothing if sampling in pesticide-treated areas.

## Method

- Enter the water, checking with the long pole that it is safe to do so and that the depth is not too great.
- Insert the sampling device and lower to the substrate to take the sediment sample. Note the depth of sediment which is being sampled with the folding ruler. (If using the Eckman grab, see method sheet on grab sampling and chapter 9.)
- Transfer the sample into a glass jar or wrap in aluminium foil and place in a polythene bag after draining away any water collected with the sample; the minimum sample size should approximately half fill a 500 ml capacity jar. Secure lid on the container; if using a bag, seal with a wire tie. At each sample collection point collect a minimum of three replicates.
- Write the sample details on the outside of the bag or jar using a permanent marker pen.
- Place the sample bag or jar inside another bag, prepare a label detailing the sample and site details and put the label into the bag. Seal the outer bag.
- Repeat the sampling process such that there is a minimum of two replicates from each identified site.
- Record the sampling details on the prepared data sheets (see page 143).
- Place the sample container into the sample box and secure it with packing material; ensure that it will not move or rattle against other containers during transportation.
- Clean the sampling tool with water and detergent, followed by acetone, between samples.



## **OTHER CONSIDERATIONS**

Sample in unsprayed areas first.

If entering the water to take a sample, use the pole to check the depth and that it is safe to proceed. Watch out for the presence of crocodiles and beware of bilharzia. Ensure that adequate protective clothing is worn when working in water.

Change gloves between sample sites to avoid cross-contamination. Seal used gloves in a labelled plastic bag, until proper disposal can be organized.

Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where possible keep the sample chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

# Sampling terrestrial vegetation for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of large, clean filter papers or clean blotting paper; manilla (paper) envelopes; cloth sachets of silica gel (kept in a sealed container before use); portable balance, 0–100 g capacity (if the samples are to be weighed in the field); disposable gloves; site map; compass; tape measure; cool-box (if available).

Thoroughly check all equipment before you go into the field.

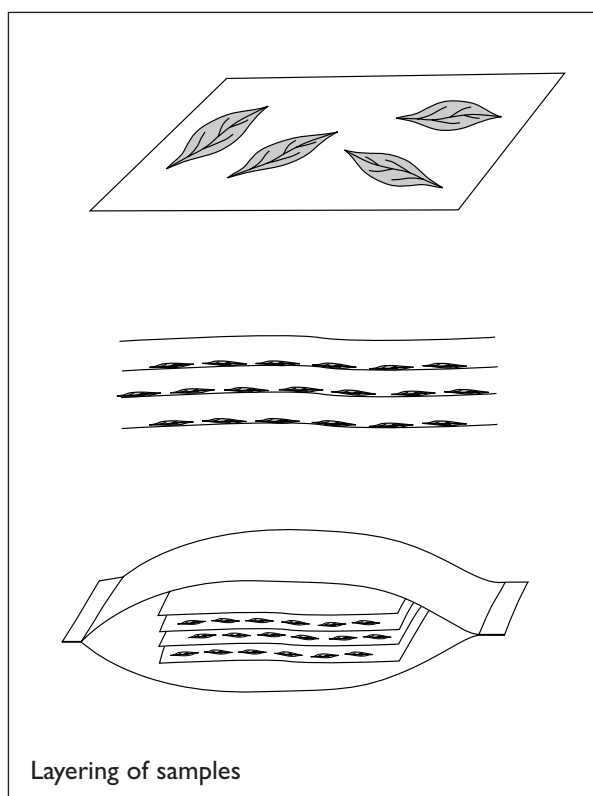
Select the sampling site and mark the site plan for future reference.

Select the vegetation to be sampled according to the agreed sampling plan; generally only grass and leaves removed from trees and shrubs will be sampled.

Wear protective clothing when sampling in pesticide-treated areas.

## Method

- Wearing disposable gloves, remove the sample (cutting required vegetation with scissors), weigh it (if necessary), note the weight and then cover the sample with filter paper or blotting paper; it is best to lay the material between sheets of paper rather than wrapping it.
- Carefully put the sample into a manilla envelope, preferably without folding.
- Write a label containing all the sample information and place the label with the sample inside the envelope.
- Copy the same information on to the outside of the envelope and on to the prepared data sheet (see page 143).
- Place a sachet of silica gel inside the envelope and close the envelope by tucking in the flap. Do not seal the envelope.
- Place the envelope into a sample box or bag, keeping the envelope horizontal where possible so that the sample is kept properly layered between the sheets of paper.
- Clean scissors with water and detergent, then wipe with acetone, before taking the next sample.



## OTHER CONSIDERATIONS

Do not sample twigs or branches.

Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where possible keep the envelopes chilled (e.g. in a refrigerator whilst the sample is awaiting transportation to the laboratory). **Do not freeze the samples** unless specific additional instructions have been received from the laboratory.

Remove gloves (keep in a labelled plastic bag until return to base for disposal) and put on a new pair before taking the next sample.

# Sampling aquatic vegetation for residues

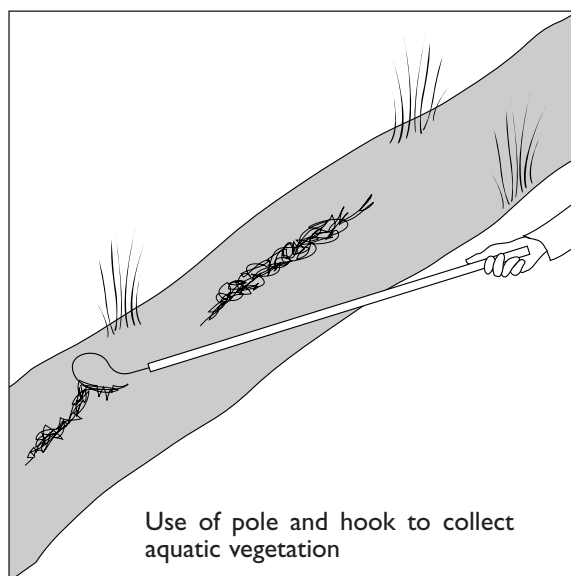
## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of clean glass jars (250 ml capacity) with aluminium foil-lined screw-caps or strong polyethylene bags 30 x 20 cm (or similar); plastic-coated wire ties for securing the bags; wellington (rubber) boots; rubber or nitrile gloves; site map; compass; tape measure; 2 m wooden pole with hook on one end; cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass vessels being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Select the sampling site and mark the site plan for future reference.  
Wear protective clothing when sampling in pesticide-treated areas.

## Method

- Where the selected vegetation can be reached from the shore take the desired sample by hand (wearing nitrile gloves) or using the pole and hook.
- Enclose the sample in clean filter paper or blotting paper to remove excess water. Where filter or blotting paper is not available, paper towels or tissue can be used. Samples of these should be provided to the analytical laboratory to check for possible co-extractives which could interfere with the analysis. Wherever possible, analytical checks on the suitability of the material should be completed before sampling commences.
- Remove the paper wrapping and place the sample into a glass jar or wrap in aluminium foil and place in a polyethylene bag; close the jar with the appropriate screw-cap or the bag with a wire tie.
- Dry the outside of the container and mark it with the sample code number.
- Place the container inside a polyethylene bag. Write a sample label containing all relevant sample details, including the sample code, and place the label inside the bag. Secure the bag with a wire tie.
- Record all sample details, including sample code, on the prepared data sheets (see page 143).
- Clean pole and hook with detergent and water, then with acetone, before taking the next sample.



## OTHER CONSIDERATIONS

If entering the water to take a sample, use the pole to check the depth and that it is safe to proceed. Watch out for the presence of crocodiles and beware of bilharzia. Ensure that adequate protective clothing is worn during sampling.

If, when entering the water to take the sample, sediment is disturbed, it is important to allow this to settle before taking the vegetation sample.

Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where possible keep the sample chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

Replace the gloves with a new, clean pair before taking the next sample. Seal used pair in a labelled plastic bag until proper disposal can be arranged.

# Sampling fish for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of clean glass jars (100–200 ml capacity) with aluminium foil-lined screw-caps; supply of polyethylene bags, 25 x 50 cm, or similar; portable balance, 0–100 g or 0–1000 g capacity (where field weighing of whole samples or of organs may be necessary); supply of dilute (8–9%) formalin solution in a clean container; rubber or nitrile gloves; disposable gloves; metal tongs or forceps; sharp knife or scalpel; cotton wool; aluminium foil; acetone (analar grade); cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass jars being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Select the sampling site and mark the site plan for future reference.  
Wear protective clothing when sampling in pesticide-treated areas.

## Method

- Capture the fish by appropriate means (see chapter 10). Samples can also be obtained from local fisherfolk as long as they are fresh and if the area of capture can be defined and time of capture assured.
- Where fish are small enough to constitute an individual sample, wrap each fish individually in aluminium foil and place within a polyethylene bag. Prepare a label including all relevant sample details and put the label inside the bag with the sample. Seal the bag with a wire tie. (See also bullet point 6.)
- Put the sample bag inside a second polyethylene bag and prepare and insert a second, identical label. Seal the bag with a wire tie.
- Record all sample details on the prepared data sheets (see page 143).
- Pack the samples into the sample box.
- Where individual body organs or muscle tissue are to be analysed, these are best removed on return to base camp. Where the size of the specimen makes this impracticable, the organs and tissue may have to be removed in the field and transported back to camp in glass sample bottles containing formalin solution. Weigh the tissue samples after removal and before placing in formalin; record the weights.
- Organs and tissue from the same specimen should be stored in separate bottles and the labelling and data sheet records should clearly show what has been done.
- The jars should be sealed and placed inside polyethylene bags, which are then sealed as a precaution against leakage. Each sample jar should contain a paper label, written in pencil, and the outer polyethylene bag should contain a second, identical label.
- Wash and rinse all sampling equipment, dissecting equipment and tools with water and rinse with acetone between samples.

## OTHER CONSIDERATIONS

If entering the water to take a sample, use the pole to check the depth and that it is safe to proceed. Watch out for the presence of crocodiles and beware of bilharzia. Ensure that adequate protective clothing is worn.

Where dissection is required, it is important to ensure that there is no risk of sample contamination. The dissection should be carried out on a clean surface covered with a material such as aluminium foil. Fresh foil should be used for each dissection. Fresh disposable gloves should be used for each specimen and any knives or forceps used cleaned with acetone between use.

Disposable gloves should be worn for the dissection work and when handling the formalin solution. The gloves should only be used once and then removed and sealed in a labelled plastic bag, until proper disposal. Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.

Where possible keep the sample chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

# Sampling birds and small mammals for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of clean glass jars (100–500 ml capacity) with aluminium foil-lined screw-caps; supply of polyethylene bags 25 x 50 cm, or similar; supply of dilute (8–9%) formalin solution in a clean container; bird capture net; net on pole or suitable traps for small mammals; rubber or nitrile gloves; disposable gloves; metal tongs or forceps; sharp knife or scalpel; portable balance, 0–100 g capacity; cotton wool; aluminium foil; acetone (analar grade); cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass jars being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Select the sampling site and mark the site plan for future reference.  
Wear protective clothing when sampling in pesticide-treated areas.

## Method

- Capture the specimen (see chapters 12 and 13) and kill by appropriate means (humanely).
- Where animals are small enough to constitute an individual sample, wrap each sample individually in aluminium foil and place within a polyethylene bag. Prepare a label including all sample details (including sex) and put the label inside the bag with the sample. Seal the bag with a wire tie.
- Put the sample bag inside a second polyethylene bag and prepare and insert a second, identical label. Seal the bag with a wire tie. (See also bullet point 7.)
- Record all sample details on the prepared data sheets (see page 143).
- Pack the samples into the sample box.
- In particularly hot climates and where chilling facilities are not immediately available, small specimens should have an incision made in the abdominal wall and the whole sample then immersed in formalin contained within a glass, screw-capped jar. The jar should be placed within a polyethylene bag which should be sealed with a wire tie. Each sample jar should contain a paper label, written in pencil, and the outer polyethylene bag should contain a second, identical label. In such cases, the sample should be weighed before immersion in formalin and the weight carefully recorded.
- Where individual body organs or muscle tissue are to be analysed, these are best removed on return to base camp. Where the size of the specimen makes this impracticable, however, and particularly in very hot climates, the organs and tissue may have to be removed in the field and transported back to camp in sample bottles containing formalin solution. The organs or body tissue sample should be weighed before immersion in formalin and the weight carefully recorded.
- Where dissection is required, it is important to ensure that there is no risk of sample contamination. The dissection should be carried out on a clean surface covered with a material such as aluminium foil. Fresh foil should be used for each dissection. Fresh disposable gloves should be used for each specimen and any knives or forceps used cleaned with acetone between use.
- Organs and tissue from the same specimen should be stored in separate bottles and the labelling and data sheet records should clearly show what has been done.
- The jars should be sealed and placed inside polyethylene bags, which are then sealed with wire ties, as a precaution against leakage.

## OTHER CONSIDERATIONS

Disposable gloves should be worn for the dissection work and when handling the formalin solution. The gloves should only be used once and then removed; fresh gloves should be worn for each sample handled (used gloves being sealed in a labelled plastic bag, until proper disposal can be organized). Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat. Where possible keep the sample chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.

# Sampling amphibians and reptiles for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details); sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of clean glass jars (100–500 ml capacity) with aluminium foil-lined screw-caps; supply of polyethylene bags large enough to contain the largest sample container; plastic-coated wire ties; supply of dilute (8–9%) formalin solution in a clean container; sample net on 1 m pole; rubber or nitrile gloves; disposable gloves; metal tongs or forceps; sharp knife or scalpel; portable balance, 0–100 g capacity; cotton wool; aluminium foil; acetone (analar grade); cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass jars being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Select the sampling site and mark the site plan for future reference.  
Wear protective clothing when sampling in pesticide-treated areas.

### Method

- Using the sample net, or by hand if appropriate (wear gloves), capture the specimen and stun (e.g. for lizards, frogs, small snakes or chelonians) with a sharp tap or finger-flick to the top of the head.
- Weigh the sample and record the weight.
- Immerse the whole specimen in formalin solution in an appropriately sized glass or aluminium container. (See also bullet point 10.)
- After 30 min remove the specimen using metal tongs or forceps (formalin solution can damage human skin) and make an incision in the abdominal wall. Return the specimen to the formalin solution.
- Prepare a suitable label, written in pencil, containing all of the relevant sample information and place in the container. Seal the container with the screw-cap, ensuring that no leaks occur.
- Label the outside of the container with all relevant sample details using a permanent marker pen.
- Place the container inside a polyethylene bag and seal with a wire tie (a precaution against leakage of the formalin).
- Record the sampling details on the prepared data sheets (see page 143).
- Pack the sample container into the sample box using packing material to ensure that the container cannot move or be damaged during transportation.
- Where individual body organs are to be analysed, these are best removed on return to base camp. Where the size of the specimen makes this impracticable, the organs may have to be removed in the field and transported back to camp in sample bottles. Organs from the same specimen should be stored in separate bottles and the labelling and data sheet records should clearly show what has been done.
- Where dissection is required, it is important to ensure that there is no risk of sample contamination. The dissection should be carried out on a clean surface covered with a material such as aluminium foil. Fresh foil should be used for each dissection. Any knives or forceps used should be cleaned with acetone between use.

## OTHER CONSIDERATIONS

Where the period of transportation is long or involves the use of aircraft, the specimen should be removed from the formalin (after a minimum of 48 h in the preservative) and wrapped in a formalin soaked square of cotton. The cotton bundle is then doubly wrapped in aluminium foil and sealed in a polythene bag. Ensure that all sample details are transferred to the new packaging. Wherever possible the original labels should be used; where this is not possible, ensure that all details are correctly transposed on to the new labels. **Double check** to prevent any error. Where the journey to the laboratory is made on good, level roads and there is no associated journey by air, the samples can remain in formalin solution.

Where dissection is required, it is important to ensure that there is no risk of sample contamination. The dissection should be carried out on a clean surface covered with a material such as aluminium foil. Fresh foil should be used for each dissection. Fresh disposable gloves should be used for each specimen and any knives or forceps used cleaned with acetone between use.

Disposable gloves should be worn for the dissection work and when handling the formalin solution. The gloves should only be used once and then removed; fresh gloves should be worn for each sample handled and used gloves sealed in a labelled plastic bag, until proper disposal can be arranged.

# Sampling invertebrates for residues

## DON'T FORGET

**EQUIPMENT:** Clip-board (carry a plastic bag in your pocket into which you can put the board if it rains); data sheets for sample site details; spare paper; sharp pencils; pens; labels; string; eraser; scissors or penknife; permanent marker pens; supply of clean glass jars (25–100 ml capacity) with perforated and unperforated aluminium foil-lined screw-caps; supply of polyethylene bags, 25 x 50 cm, or similar; supply of dilute (8–9%) formalin solution in a clean container; disposable gloves; metal tongs or forceps; sharp knife or scalpel; cotton wool; cool-box where available or a strong sample box to contain the sample vessels with appropriate packing (cardboard or foam rubber) to prevent glass jars being damaged or broken during transit.

Thoroughly check all equipment before you go into the field.  
Select the sampling site and mark the site plan for future reference.  
Wear protective clothing when collecting samples in pesticide-treated areas.

## Method

- Capture the specimen by appropriate means (see chapter 8) and transfer to a suitable size aluminium container.
- Prepare a label including all sample details and put the label inside the canister with the sample. Fit the appropriate screw-cap (perforated for live insect samples, to allow ventilation or without perforation if the specimens are to be preserved in formalin or kept dry and frozen. Label the outside of the canister with the appropriate sample details or sample code.
- Where the sample is preserved in formalin, place the sample container inside a polyethylene bag and prepare and insert a second, identical label. Seal the bag with a wire tie.
- Record all sample details on the prepared data sheets (see page 143).
- Pack the samples into the sample box.
- In particularly hot climates, large specimens should have an incision made in the abdominal wall and the whole sample then immersed in formalin contained within a screw-capped jar. The jar should be placed within a polyethylene bag which should be sealed with a wire tie. Each sample jar should contain a paper label, written in pencil, and the outer polyethylene bag should contain a second, identical label.
- Disposable gloves should be worn when handling the formalin solution. The gloves should only be used once and then removed; fresh gloves should be worn for each sample handled.
- Clean any equipment used to sample or handle the invertebrates between samples, using detergent and water, rinsing thoroughly in clean water and finally in acetone.

## OTHER CONSIDERATIONS

Always collect samples in unsprayed areas first; pesticide-treated areas later.  
Where a cool-box is available, it should be used to transport the samples whilst in the field. On no account should the collected samples be exposed to direct sunlight or extremes of heat.  
Where possible, keep the samples chilled (e.g. in a refrigerator) whilst awaiting transportation to the analytical laboratory.  
Used disposable gloves should be sealed in a labelled plastic bag, until proper disposal can be organized.