Water Quality Monitoring - A Practical Guide to the Design and Implementation of Freshwater Quality Studies and Monitoring Programmes
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Chapter 5 - FIELD WORK AND SAMPLING

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The field work associated with the collection and transport of samples will account for a substantial proportion of the total cost of a monitoring programme. Sampling expeditions should, therefore, be planned and carried out in such a way that efforts are not wasted. If, for example, an essential piece of equipment is forgotten or an inadequately described sampling station cannot be found, the value of that particular sampling expedition is seriously compromised. Similarly, if unrealistic estimates of travel time are made and an expedition takes longer than intended, samples may be held longer than the maximum allowable storage time and the results of analyses will be of questionable value.

Special aspects of field work and sampling associated with biological methods and sediment measurements are given in Chapters 11 and 13 and in the companion guidebook *Water Quality Assessments*.

The sample collection process should be co-ordinated with the laboratory so that analysts know how many samples will be arriving, the approximate time of their arrival and the analyses that are to be carried out, and can thus have appropriate quantities of reagent chemicals prepared.

It is good practice to prepare a checklist such as the one on the following page, so that nothing is missing or forgotten before a sampling expedition is undertaken. Many of the items in the list are self-explanatory; others are described more fully in later sections of this chapter.

Personnel who will collect water, biota or sediment samples must be fully trained in both sampling techniques and field test procedures. They should also be aware of the objectives of the monitoring programme since these will have some influence on the sampling procedures. Obtaining a sample that is fully representative of the whole water body is difficult and the collection and handling of samples are also frequent sources of error (often greater errors than those arising during analysis). Thus the choice of a representative sampling point and the use of appropriate sampling techniques are of fundamental importance (see Chapter 3 and the companion guidebook *Water Quality Assessments*).

Checklist for preparing for field work

Paperwork

√ Itinerary
√ Inventory details of sampling stations; maps

√ List of samples required at each sampling station
√ List of stations where water level readings are to be recorded (see Chapter 12)
Co-ordination
√ Local co-ordination, for example, to ensure access to sites on restricted or private land

√ Institutional co-ordination, for example, for travel arrangements or sample transport

√ Notify laboratories of expected date and time of sample arrival

√ Check any available sources of information on local weather conditions and feasibility of travel

For sampling
√ Sample bottles, preservatives, labels and marker pens
√ Sample storage/transit containers and ice packs
√ Filtering apparatus (if required)
√ Samplers/sampling equipment
√ Rubber boots, waders, etc.
√ Standard operating procedures for sampling
√ Spares of all above items if possible and when appropriate
For documentation
√ Pens/wax crayons
√ Sample labels
√ Field notebook
√ Report forms
For on-site testing
√ List of analyses to be performed on site

√ Check stocks of consumables (including distilled water, pH buffers, standards and blanks); replenish and refresh as appropriate

√ Check and calibrate meters (pH, conductivity, dissolved oxygen, turbidity, thermometers)

√ Other testing equipment according to local practice

√ Standard operating procedures and equipment manuals

√ Spares (e.g. batteries)

Safety
√ First-aid kit
√ Waders, gloves, etc.
√ Fire extinguisher (if appropriate)
Transport
√ Does assigned vehicle have sufficient capacity for personnel, supplies and equipment?
√ Is vehicle road-worthy? Check battery, lubrication, coolant, windshield washer
√ Is there sufficient fuel for the trip, either in the tank, in fuel cans, or available en route?
√ Is the spare tyre inflated, is there a jack, wheel wrench and tool kit?
Double-check
√ When was equipment last calibrated?

√ Itinerary against travel details on inventory

√ Accessories for equipment and meters (including cables, chargers and spare batteries) and consumables

Only if samples can be taken consistently from the same locations, can changes in the concentration of water quality variables with time be interpreted with confidence. It is advisable to carry out a pilot programme before the routine monitoring programme begins (see section 3.4). This can be used as a training exercise for new personnel and will provide the opportunity to make a final selection of sampling stations on the basis of whether they are representative of the whole water body as well as readily accessible.

Programme managers and laboratory personnel should accompany field personnel on field expeditions from time to time. This provides opportunities for field supervision as part of in-service training and for everyone working on the programme to appreciate the problems and needs of field work.

On-site testing is common for certain variables, especially those that may change (physical, chemical or biological) during transport. Dissolved oxygen, turbidity, transparency, conductivity, pH, temperature and, to a lesser extent, thermotolerant (faecal) and total coliform counts are the variables most often measured on site. Procedures for carrying out analyses in the field are covered in Chapters 6 and 10.

The special problems of analytical quality control during field testing or testing using portable equipment are outlined in section 6.7, and further information on minimising the risks associated with this approach is provided in Chapter 9.

5.1 Sample containers

Containers for the transportation of samples are best provided by the laboratory. This ensures that large enough samples are obtained for the planned analyses and that sample bottles have been properly prepared, including the addition of stabilizing preservatives when necessary. It is essential to have enough containers to hold the samples collected during a sampling expedition. Sample containers should be used only for water samples and never for the storage of chemicals or other liquids. Glass containers are commonly used and are appropriate for samples for many analyses, but plastic containers are preferred for samples intended for certain chemical analyses or for biota or sediments. Plastic has the obvious advantage that it is less likely to break than glass.

Sample containers must be scrupulously clean so that they do not contaminate the samples placed in them. Table 5.1 provides general information on appropriate types of sample container and the recommended procedures for cleaning them when water samples are to be used for chemical analysis.

For microbiological analysis, strong, thick-walled, glass sample bottles with a minimum capacity of 300 ml should be used. They should have screw caps of a type that will maintain an effective seal, even after they have been sterilised many times in an autoclave. Some technicians fasten a Kraft paper cover over the bottle caps before autoclaving to protect them from contamination during handling. Alternatively, plastic or aluminium sleeves may be used. The neck of the bottle should not be plugged with cotton wool. To prepare sample bottles, they should be washed with a non-ionic detergent and rinsed at least three times (five is better) with distilled or deionised water before autoclaving. New bottles require the same preparation. If distilled or deionised water is not available, clean chlorine-free water may be used.

If chlorinated water is being collected for microbiological analysis, sufficient sodium thiosulphate should be added to the sample bottles to neutralise the chlorine. The recommended amount is 0.1 ml of a 1.8 per cent solution of sodium thiosulphate for each 100 ml of sample bottle volume; this should be added to the bottles before autoclaving. Sample bottles should not be rinsed with sample water or allowed to overflow because this would remove the dechlorinating chemical.

Table 5.1 Sample containers and their recommended washing procedures for selected water quality variables

Variable(s) to be analysed	Recommended container[1]	Washing procedure
Organochlorinated pesticides and PCBs Organophosphorus	1,000 ml glass (amber) with teflon-lined cap	Rinse three times with tap water, once with chromic acid[2], three times with organic-free water, twice with washing acetone, once with special grade[3] acetone, twice with pesticide grade hexane and dry (uncapped) in a hot air oven at 360 °C
Pentachlorophenol Phenolics Phenoxy acid herbicides	1,000 ml glass (amber) with teflon-lined cap	Rinse three times with tap water, once with chromic acid[2], three times with organic-free water, twice with washing acetone, once with special grade[3] acetone, twice with pesticide grade hexane and dry (uncapped) in a hot air oven at 360 °C for at least 1 h
Aluminium, Antimony, Barium, Beryllium, Cadmium, Chromium[4], Cobalt, Copper, Iron, Lead, Lithium, Manganese, Molybdenum, Nickel, Selenium, Strontium, Vanadium, Zinc	500-1,000 ml polyethylene (depending upon number of metals to be determined)	Rinse three times with tap water, once with chromic acid[2], three times with tap water, once with 1:1 nitric acid and then three times with ultrapure distilled water[5] in that order
Silver	250 ml polyethylene (amber)	Rinse three times with tap water, once with chromic acid[2], three times with tap water, once with 1:1 nitric acid and then three times with ultrapure distilled water[5] in that order
Mercury	100 ml glass (Sovirel)	Rinse three times with tap water, once with chromic acid[2], three times with tap water, once with 1:1 nitric acid and then three times with ultrapure distilled water[5] in that order
Acidity, Alkalinity, Arsenic, Calcium, Chloride, Colour, Fluoride, Hardness, Magnesium, Non- filterable residue, pH, Potassium, Sodium, Specific conductance, Sulphate, Turbidity	1,000 ml polyethylene	Rinse three times with tap water, once with chromic acid[2], three times with tap water, once with 1:1 nitric acid and then three times with distilled water in that order
Carbon, total organic Nitrogen: ammonia Nitrogen: nitrate, nitrite Nitrogen: total	250 ml polyethylene	Rinse three times with tap water, once with chromic acid[2], three times with tap water, and three times with distilled water, in that order
Phosphorus, total	50 ml glass (Sovirel)	Rinse three times with tap water, once with chromic acid[2], three times with tap water, and three times with distilled water, in that order

[1] Teflon containers can also be used to replace either the recommended polyethylene or glass containers
[2] Chromic acid - 35 ml saturated $Na_2Cr_2O_7$ per litre reagent grade conc. H_2SO_4

[3] Special grade acetone - pesticide grade when GC analysis to be performed, UV grade for LC analysis
[4] Chromic acid should not be used when the sample will be analysed for chromium
[5] Ultrapure distilled water is obtained by passing distilled water through a Corning model AG-11 all-glass distillation unit and then through a Millipore Super Q Ultrapure Water System containing a prefilter cartridge, an activated carbon cartridge and a mixed bed deionisation cartridge
Source: After WMO, 1988

Some water quality variables are unstable and, unless an analysis can be carried out immediately after the sample is obtained, it is necessary to stabilize the sample by adding a chemical preservative. It is often convenient to add chemical preservatives to containers in the laboratory rather than in the field. When this is done, it is essential that the containers be clearly labeled with the name, concentration and quantity of the preservative chemical, the volume of the sample to be collected and the variables for which the sample is to be analyzed. If preservatives are not added to containers in the laboratory, the chemicals, pipettes and directions for adding preservatives must be included in the kit of supplies and equipment taken on the sampling expedition. The subject of sample preservation is dealt with more fully in section 5.6.

5.2 Types of sample

5.2.1 Sampling surface waters

Two different types of sample can be taken from rivers, lakes and similar surface waters. The simplest, a “grab” sample, is taken at a selected location, depth and time. Normally, the quantity of water taken is sufficient for all the physical and chemical analyses that will be done on the sample. Sometimes, if the sampler is small and many analyses are to be done, two grab samples will be taken at the station and will be mixed in the same transport container. Grab samples are also known as “spot” or “snap” samples.

Composite or integrated samples, i.e. samples made up of several different parts, are often needed to fulfil some specific monitoring objectives. Composite samples may be of the following types:

- *Depth-integrated:* most commonly made up of two or more equal parts collected at predetermined depth intervals between the surface and the bottom. A piece of flexible plastic piping of several metres in length, and which is weighted at the bottom, provides a simple mechanism for collecting and integrating a water sample from the surface to the required depth in a lake. The upper end is closed before hauling up the lower (open) end by means of an attached rope. Integrated samples can also be obtained using a water pump (submersible pumps are available which allow sampling at depth) which is operated at a steady pumping rate while the water inlet is drawn upwards between the desired depths at a uniform speed.

- *Area-integrated:* made by combining a series of samples taken at various sampling points spatially distributed in the water body (but usually all at one depth or at predetermined depth intervals).

- *Time-integrated:* made by mixing equal volumes of water collected at a sampling station at regular time intervals.

- *Discharge-integrated:* It is first necessary to collect samples and to measure the rate of discharge at regular intervals over the period of interest. A common arrangement is to sample every 2 hours over a 24-hour period. The composite sample is then made by mixing portions of the individual sample that are proportional to the rate of discharge at the time the sample was taken.

5.2.2 Sampling groundwater

Groundwater samples are normally obtained from existing drilled wells, dug (shallow) wells or springs. Occasionally, during the course of a hydro-geological survey, test wells may be drilled and these can be used for monitoring purposes. The usual situation, however, is that a producing well or spring will be a groundwater quality monitoring station.

If the groundwater source is a flowing spring or a well equipped with a pump, the sample can be obtained at the point of discharge. The water should flow for several minutes before sampling until it has reached constant conductivity or temperature in order to avoid any water resident in the system's piping being taken as a sample (the piping material may have contaminated the water). Samples for dissolved oxygen analysis should be taken by inserting one end of a plastic tube into the discharge pipe and the other end into a sample bottle. The water should be allowed to flow into the bottle for sufficient time to displace the contents of the bottle at least three times. Care should be taken to ensure that no air bubbles are introduced to the sample while the bottle is being filled, since this could alter the dissolved oxygen concentration.

Special care must be taken when sampling from springs that do not have an overflow and from shallow wells without pumps. The sampling container must not be allowed to touch the bottom of the well or spring catchment since this would cause settled particles to become resuspended and to contaminate the sample. Sometimes, a spring catchment is higher than the surrounding ground and this permits water to be siphoned into the sample bottle. If this is done, water should be allowed to run through the hose for 2 - 3 minutes to rinse it thoroughly before the sample is collected. Siphoned samples are suitable for dissolved oxygen determination provided that the sample bottle is allowed to overflow a volume of at least three times its capacity.

The depth within an aquifer from which a sample of water is collected from a well is determined by the location of the well screen (as described in section 2.2) and cannot be varied by the collector, because water enters a well at the level of the screen. Similarly, water enters a spring through fissures in the rock. Consequently, a groundwater sample can only be obtained as a grab sample. The greatest danger of getting a non-representative sample occurs when insufficient water has been pumped before the sample is collected and that the sample obtained is representative of the well rather than of the aquifer.

5.3 Water samplers

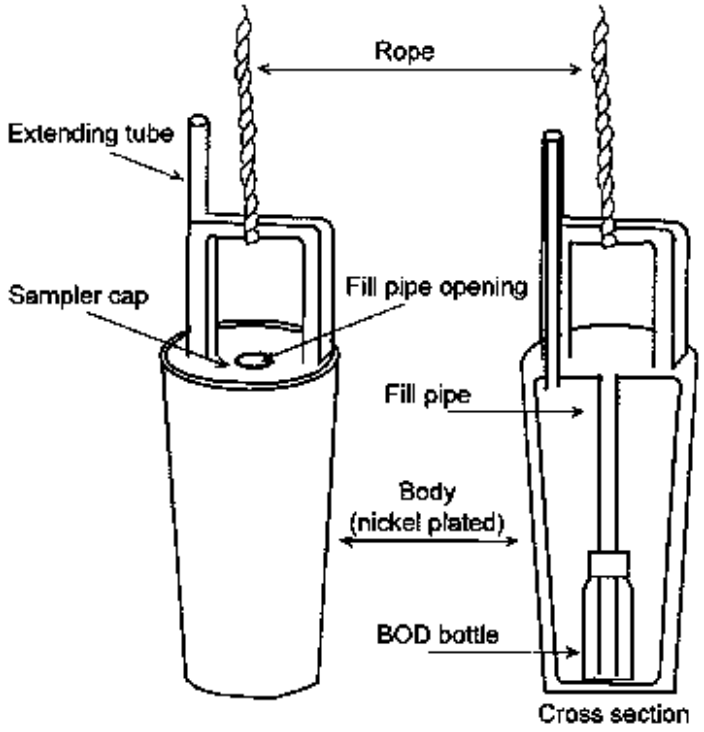
Several different types of sampler are available, many of them designed for specific purposes. The three types described here are those that are most useful for a general water sampling programme. Equipment required to obtain samples for biological and sediment analysis is described in the relevant sections of Chapters 11 and 13.

5.3.1 Dissolved oxygen sampler

A dissolved oxygen sampler is a metal tube about 10 cm in diameter and 30 cm in length, sealed at one end and with a removable cap (usually threaded) at the other. A bracket is located inside the tube in such a way that a 300-ml BOD bottle can be placed in the bracket with the top of the bottle 2 - 3 cm below the top of the sampler. The sampler cap has a tube extending from its underside down into the BOD bottle when the cap is in place. The upper end of this tube is open and flush with the outside face of the sampler top. A second tube in the sampler cap is flush with the inside face and extends upwards for about 8 - 10 cm. This second tube is sometimes incorporated into the frame to which the lowering rope is fastened. Figure 5.1 shows a typical dissolved oxygen sampler.

When the sampler is used, a BOD bottle is placed in the bracket, the sampler cap is fitted in place, and a lowering rope is fastened to the sampler which is then lowered vertically to the depth from which the sample is to be taken. Air in the sampler flows out through the highest tube and, consequently, water enters the BOD bottle through the lower tube. The volume of the sampler is about five times the volume of the bottle, therefore the incoming water flushes out the bottle at least four times and the water that finally remains in the bottle will have had no contact with the air that was originally in the sampler. Provided that the sampler is lowered quickly to the desired sampling depth, the sample obtained should be representative, in terms of its dissolved oxygen content. If a sample needs to be taken from great depth, inflow to the sampler can be prevented with a cork or similar device that can be removed when the desired depth is reached.

Figure 5.1 Dissolved oxygen sampler (Adapted from WMO, 1988)



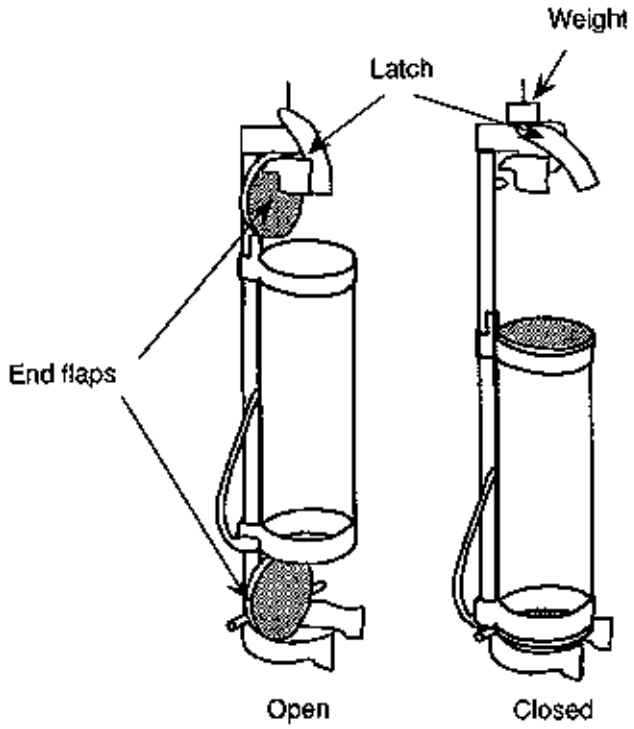
When the sampler is returned to the surface, the cap is removed and a ground-glass stopper is placed in the (ground-glass) neck of the BOD bottle before it is taken out of the sampler. Further handling of the dissolved oxygen sample is described in section 6.5. The water remaining in the sampler can be used for other analyses but it must be remembered that this will be water that flowed into the sampler between the surface and the depth at which the dissolved oxygen sample was taken. Moreover, its volume will be only about 1.5 litres, which may not be enough for all of the intended analyses.

5.3.2 Depth sampler

The depth sampler, which is sometimes called a grab sampler, is designed in such a way that it can retrieve a sample from any predetermined depth. A typical depth sampler is shown in Figure 5.2. It consists of a tube, approximately 10 cm in diameter and 30 cm in length, fastened to a frame along which it can slide. The frame has projections at each end so that the tube can not slide off. The ends of the tube are covered by spring-loaded flaps, which can be held in the fully open position by latches. The latches can be released by applying a small amount of pressure to a lever. To accomplish this, a weight (called a “messenger”) is dropped down the lowering rope, the latch is tripped and the ends of the tube close.

When the sampler is in use, the end flaps are latched into the open position. As the sampler is lowered to the required depth with the lowering rope, water passes through the open ends so that, at any depth, the water in the sampler is the water from that depth. When the desired depth is reached, the messenger weight is dropped down the rope, the latch is tripped and the end flaps close. The sampler is brought to the surface and its contents are transferred to a sample bottle. A sample obtained in this way can be used for all chemical analyses except dissolved oxygen. A simpler and less expensive model of depth sampler, suitable for moderate depths (< 30 m) is illustrated in Figure 5.3.

Figure 5.2 Depth sampler



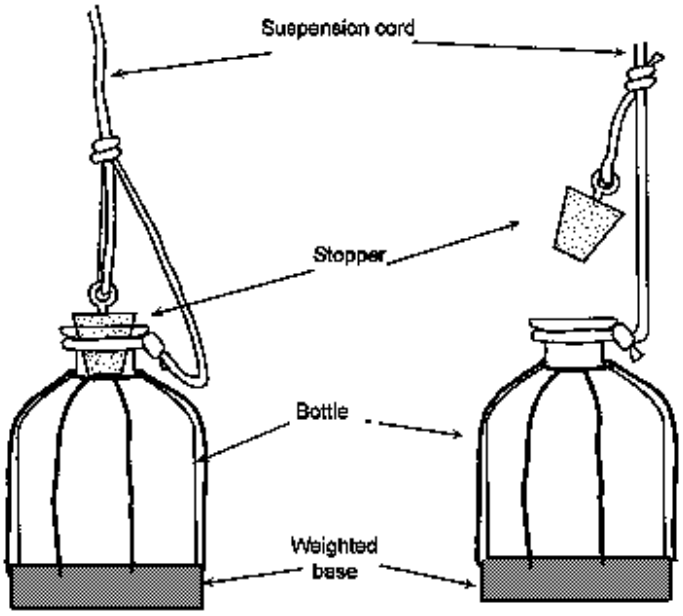
5.3.3 Multi-purpose sampler

A multi-purpose sampler, also called a sampling iron, is most frequently used for taking samples in flowing streams or rivers. It consists of a weighted platform equipped with clamps or similar means of holding a sample bottle, a rudder to maintain its position in the flowing water, and rings at the top and bottom to which lowering ropes can be attached as shown in Figure 5.4. One end of the rope may be attached to the top ring and a friction release device, connected between the rope and the bottom ring, holds the bottle in an inverted position during lowering. An alternative arrangement is to use two ropes, one fastened to the lower ring and one to the upper. Both arrangements permit the collection of samples from a deep location by allowing the sampler to be lowered in an inverted position and then restored to the upright position when the required depth is reached.

The multi-purpose sampler is very easy to use for sampling near the surface. It is simply immersed in the water and allowed to fill up. For samples from greater depths, it must be lowered in the inverted position and then, when the desired depth is reached, righted either by a sharp tug on the rope (for the one-rope configuration) or by transferring restraint to the rope connected to the upper ring. Although some water may enter the sampler during its descent, this type of sampler has the advantage that the sample does not need to be transferred to another container for shipment because it can remain in the container in which

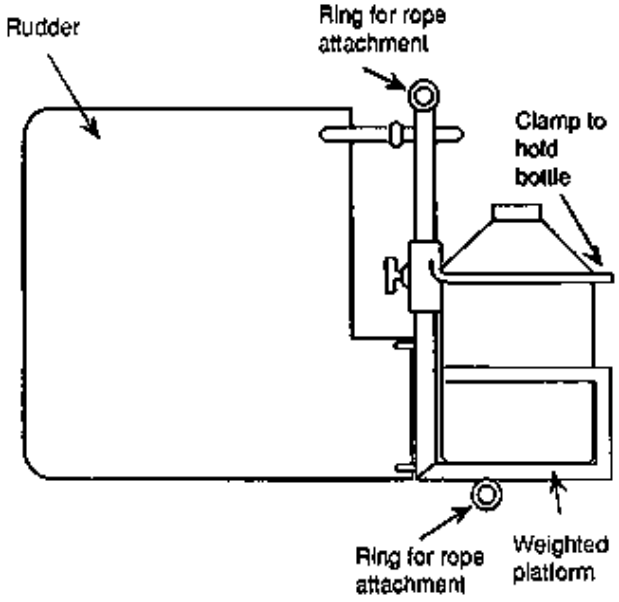
it was collected. Samples taken with the multi- purpose sampler cannot be used for dissolved oxygen determination.

Figure 5.3 Depth sampler suitable for moderate depths



When samples are taken for chemical and physical analysis from rivers and lakes, it is often sufficient merely to immerse an open-mouthed vessel, such as a bucket, below the water surface. The contents can then be poured into an appropriate set of sample bottles. Alternatively, the sample bottle can be immersed in the water and allowed to fill up. Care should be taken to avoid the entry of water from the surface since this will often contain very fine floating material that cannot be easily seen. If the water is flowing, the open mouth of the bottle should point upstream.

Figure 5.4 Multi-purpose sampler (Adapted from WMO, 1988)



5.4 Manual sampling procedures

This section deals principally with sampling for water analysis or bacteriological analysis. Details of procedures for other biological methods and for sediment measurements are given in Chapters 11 and 13 respectively.

5.4.1 Guidelines

Samples for physical and chemical analyses

The minimum sample size varies widely depending on the range of variables to be considered and the analytical methods to be employed, but it is commonly between 1 and 5 litres. The volumes required for individual analyses are summarised in Table 5.2.

Table 5.2 Sample volumes required for individual physico-chemical analyses

Analysis	Sample volume (ml)	Analysis	Sample volume (ml)
Alkalinity	100	Kjeldahl nitrogen	400
Aluminium	25	Nitrate nitrogen	200
BOD	1,000	Nitrite nitrogen	50
Boron	1	Phosphorus	100
Calcium	50	Potassium	100
Chloride	100	Selenium	1,000
Fluoride	50	Silica	50
Iron	50	Sodium	100
Magnesium	75	Sulphate	200
Manganese	90	TOC	200
Ammonia nitrogen	400	TSS	1,000

BOD Biochemical oxygen demand
TOC Total organic carbon
TSS Total suspended solids

The following general guidelines can be applied to the collection of water samples (to be analysed for physical or chemical variables) from rivers and streams, lakes or reservoirs and groundwater.

- Before collecting any sample, make sure that you are at the right place. This can be determined by the description of the station, from the position of landmarks and, in lakes, by checking the depth. If samples must be taken from a boat, a sampling station may be marked by placing a buoy at the desired location; otherwise it is necessary to identify the sampling station by the intersection of lines between landmarks on the shore.

- Do not include large, non-homogeneous pieces of detritus, such as leaves, in the sample. Avoid touching and disturbing the bottom of a water body when taking a depth sample, because this will cause particles to become suspended. As an example, the GEMS/WATER monitoring programme sets the upper size limit of particulate matter at 0.063 mm. To remove larger material pass the water sample through a sieve and collect it in a bottle for transport.

- Sampling depth is measured from the water surface to the middle of the sampler.

- Samples taken to describe the vertical profile should be taken in a sequence that starts at the surface and finishes at the bottom. When taking the sample at the maximum depth it is important to ensure that the bottom of the sampler is at least 1 m above the bottom.

- Do not lower a depth sampler too rapidly. Let it remain at the required depth for about 15 seconds before releasing the messenger (or whatever other device closes the sampler). The lowering rope should be vertical at the time of sampling. In flowing water, however, this will not be possible and the additional lowering necessary to reach the required depth should be calculated.

- A bottle that is to be used for transport or storage of the sample should be rinsed three times with portions of the sample before being filled. This does not apply, however, if the storage/transport bottle already contains a preservative chemical.

- The temperature of the sample should be measured and recorded immediately after the sample is taken.

- The sample to be used for dissolved oxygen determination should be prepared immediately after the temperature is measured. If an electronic technique is being used, a portion of the sample is carefully poured into a beaker for measurement. If the Winkler method is being used, the chemical reagents are added to the bottle in accordance with the directions contained in section 6.5.

- Separate portions of the sample should be set aside for pH and conductivity determinations. The same portion must not be used for both determinations because of the possibility of potassium chloride diffusing from the pH probe.

- At any time that the sample bottles are not closed, their tops must be kept in a clean place.

- A small air space should be left in the sample bottle to allow the sample to be mixed before analysis.

- All measurements taken in the field must be recorded in the field notebook before leaving the sampling station.

- All supporting information should be recorded in the field notebook before leaving the sampling station. Such conditions as the ambient air temperature, the weather, the presence of dead fish floating in the water or of oil slicks, growth of algae, or any unusual sights or smells should be noted, no matter how trivial they may seem at the time. These notes and observations will be of great help when interpreting analytical results.

- Samples should be transferred to sample bottles immediately after collection if they are to be transported. If analysis is to be carried out in the field, it should be started as soon as possible.

Samples for bacteriological analysis

Most of the guidelines for sampling for physical and chemical analyses apply equally to the collection of samples for bacteriological analyses. Additional considerations are:

- Samples for bacteriological analyses should be taken in a sterile sampling cup and should be obtained before samples for other analyses.

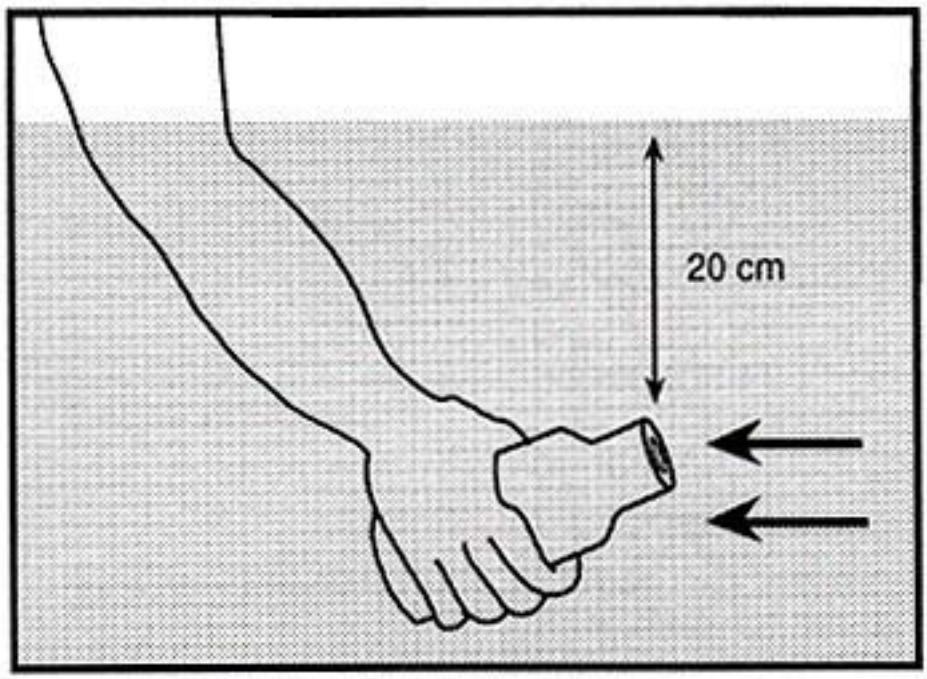
- Care must be exercised to prevent contamination of the inside of the sampling cup and sampling containers by touching with the fingers or any non-sterile tools or other objects.

- Bottles in which samples for bacteriological analyses are to be collected (or transported) should be reserved exclusively for that purpose.

5.4.2 Procedures

Sampling from a tap or pump outlet

Figure 5.5 Collecting a sample from surface water

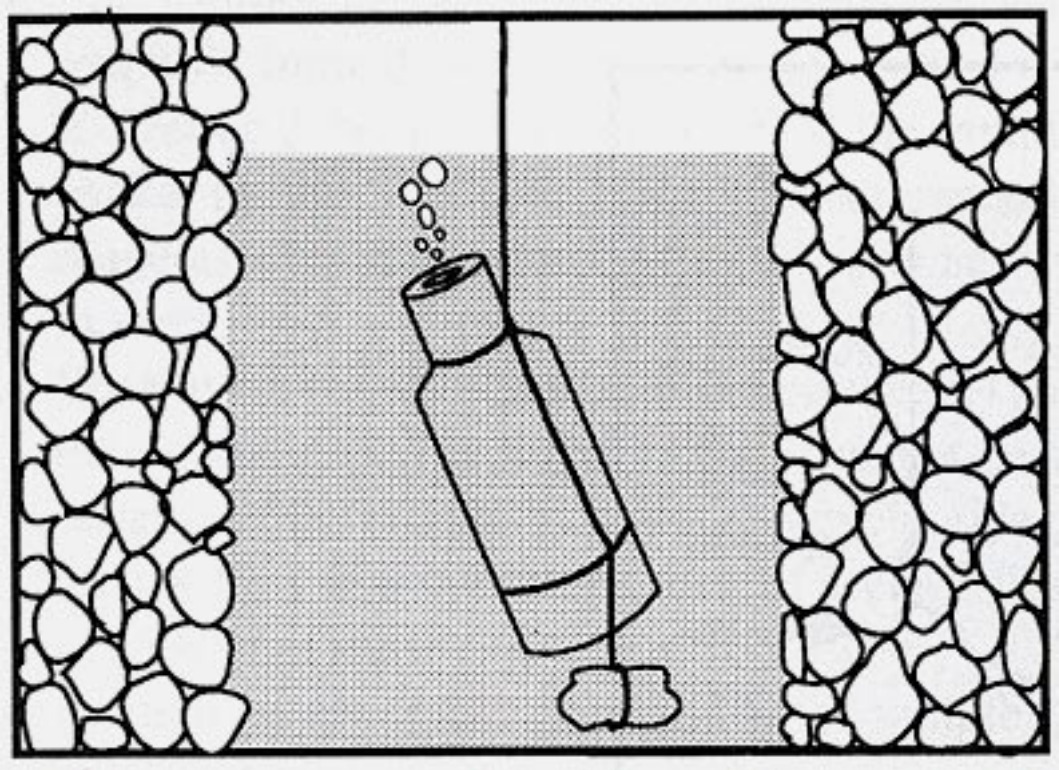


1. Clean the tap. Remove any attachments that may cause splashing from the tap. These attachments are a frequent source of contamination that may influence the perceived quality of the water supply. Use a clean cloth to wipe the outlet and to remove any dirt.
2. Open the tap. Turn on the tap to maximum flow and let the water run for 1-2 minutes. Turn off the tap.
Note: Some people omit the next two steps and take the samples at this stage, in which case the tap should not be adjusted or turned off, but left to run at maximum flow.
3. Sterilize the tap for 1 minute with a flame (from a gas burner, cigarette lighter or an alcohol-soaked cotton wool swab).
4. Open the tap before sampling. Carefully turn on the tap and allow water to flow at medium rate for 1 - 2 minutes. Do not adjust the flow after it has been set.

5. Fill the bottle. Carefully remove the cap and protective cover from the bottle, taking care to prevent entry of dust that may contaminate the sample. Hold the bottle immediately under the water jet to fill it. A small air space should be left to allow mixing before analysis. Replace the bottle cap.

Sampling water from a water-course or reservoir

Figure 5.6 Lowering a weighted bottle into a well



Open the sterilised bottle as described in step 5 above.

1. Fill the bottle (see Figure 5.5). Hold the bottle near its bottom and submerge it to a depth of about 20 cm, with the mouth facing slightly downwards. If there is a current, the bottle mouth should face towards the current. Turn the bottle upright to fill it. Replace the bottle cap.

Sampling from dug wells and similar sources

1. Prepare the bottle. With a length of string, attach a weight to the sterilised sample bottle (Figure 5.6).

2. Attach the bottle to the string. Take a 20 m length of string, rolled around a stick, and tie it to the bottle string. Open the bottle as described above.

3. Lower the bottle. Lower the weighted bottle into the well, unwinding the string slowly. Do not allow the bottle to touch the sides of the well (see Figure 5.6).

4. Fill the bottle. Immerse the bottle completely in the water and continue to lower it to some distance below the surface (see Figure 5.6). Do not allow the bottle to touch the bottom of the well or disturb any sediment.

5. Raise the bottle. Once the bottle is judged to be full, bring it up by rewinding the string around the stick. If the bottle is completely full, discard a little water to provide an air space. Cap the bottle as described previously.

5.5 Recording field observations

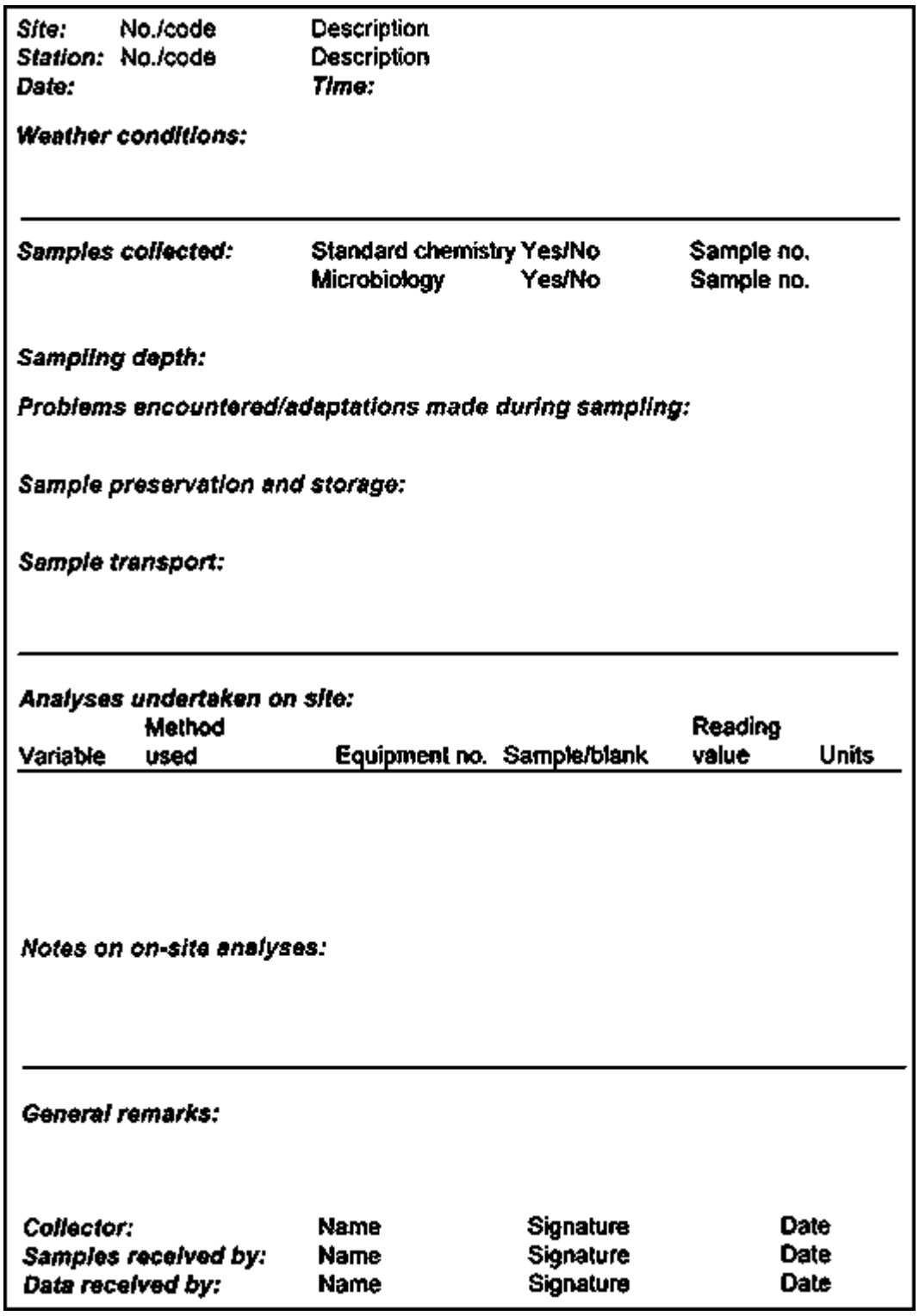
Sampling officers should have a field notebook in which all details of relevance are recorded at the time. The field book should be hard-bound and not loose-leaf. Full books should not be discarded but stored for future reference because they represent data in original form and are sometimes invaluable for reference purposes.

Details recorded should include: those noted on the sample bottle (see section 5.7), what samples were collected, and what measurements were made, how they were made, and the results obtained (including blanks, standards, etc., and the units employed).

All supporting information (any unusual local features at the site and time of sampling) should also be noted. If there has been any variation from the agreed sampling station, this should be noted, with reasons. Any need for a permanent change in sampling station should be brought to the attention of the programme co-ordinator and the inventory should be changed if necessary.

If a standard field record layout is used in place of a plain notebook, adequate space should be available for comments and observations. To facilitate field work, the layout and content of the pages should reflect the sequence in which the various procedures will be carried out (see Figure 5.7).

Figure 5.7 Example page from a field notebook



Site: No./code Description
Station: No./code Description
Date: *Time:*

Weather conditions:

Samples collected: Standard chemistry Yes/No Sample no.
Microbiology Yes/No Sample no.

Sampling depth:

Problems encountered/adaptations made during sampling:

Sample preservation and storage:

Sample transport:

Analyses undertaken on site:

Variable	Method used	Equipment no.	Sample/blank	Reading value	Units

Notes on on-site analyses:

General remarks:

Collector: Name Signature Date
Samples received by: Name Signature Date
Data received by: Name Signature Date

5.6 Sample preservation

There is little clear consensus on the best means of preserving samples for specific analyses, the practicality of preservation in the field and the length of time for which samples may be stored without deteriorating. In general, sample bottles should be resealed and stored in a clean, cool, dark environment and protected from recontamination. Additional

methods of preservation include freezing, solvent extraction and the addition of chemical preservatives.

Storage may vary according to the method employed for analyses and correct information may, therefore, be available in standard methods. If not, stability testing may have to be carried out before sampling is initiated and should, in any case, be done as part of method validation.

Suggested chemical preservatives and recommended maximum storage times for samples for various analyses are summarised in Table 5.3.

The preservative treatment may be applied immediately on sampling or may already be contained in the sample bottle. For neutralising chlorine in samples for microbiological analysis, however, sodium thiosulphate must be in the sample bottles before they are autoclaved. Where pretreatment by filtration is required, the preservative should be added immediately after filtration, as some chemicals may alter the physical characteristics of the sample. Acidification, for example, may lead to dissolution of colloidal and particulate metals.

5.7 Transportation and storage of samples

The sample collection process should be co-ordinated with the laboratory. Analysts need to know how many samples will be arriving, the approximate time of arrival and the analyses that are to be carried out, so that appropriate quantities of reagent chemicals can be prepared. If sample bottles are provided by the laboratory, this ensures that they are of adequate volume and have been properly prepared (with added chemical preservatives where necessary).

Each sample bottle must be provided with an identification label on which the following information is legibly and indelibly written:

- Name of the study.
- Sample station identification and/or number.
- Sampling depth.
- Date and time of sampling.
- Name of the individual who collected the sample.
- Brief details of weather and any unusual conditions prevailing at the time of sampling.
- Record of any stabilising preservative treatment.
- Results of any measurements completed in the field.

Table 5.3 Suggested preservative treatments and maximum permissible storage times

Variable	Recommended container[1]	Preservative	Max. permissible storage time
Alkalinity	Polyethylene	Cool 4 °C	24 h
Aluminium	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
Arsenic	Polyethylene	Cool 4 °C	6 months
BOD	Polyethylene	Cool 4 °C	4h
Boron	Polyethylene	Cool 4 °C	6 months
Cadmium	Polyethylene	2 ml Conc. HNO_3 l^{-1} sample	6 months
Calcium	Polyethylene	Cool 4 °C	7 days
Carbamate pesticides	Glass	H_2SO_4 to pH < 4, 10g Na_2SO_4 l^{-1}	Extract immediately
Carbon			
inorganic/organic	Polyethylene	Cool 4 °C	24 h
particulate	Plastic Petri dish	Filter using GF/C filter; Cool, 4 °C	6 months
Chloride	Polyethylene	Cool 4 °C	7 days
Chlorinated hydrocarbon	Glass	Cool 4 °C	Extract immediately
Chlorophyll	Plastic Petri dish	Filter on GF/C filter; freeze -20 °C	7 days
Chromium	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
COD	Polyethylene	Cool 4 °C	24 h
Copper	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
Dissolved oxygen (Winkler)	Glass	Fix on site	6h
Fluoride	Polyethylene	Cool 4 °C	7 days
Iron	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
Lead	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
Magnesium	Polyethylene	Cool 4 °C	7 days
Manganese	Polyethylene	2 ml Conc. HNO_3 l^{-1} sample	6 months
Mercury	Glass or teflon	1 ml Conc. H_2SO_4 + 1 ml 5% $2Cr_2O_7$	1 month
Nickel	Polyethylene	2 ml Conc. HNO_2 l^{-1} sample	6 months
Nitrogen			
Ammonia	Polyethylene	Cool 4 °C, 2 ml 40% H_2SO_4 l^{-1}	24 h
Kjeldahl	Polyethylene	Cool 4 °C	24 h
Nitrate + Nitrite	Polyethylene	Cool 4 °C	24 h
Organic nitrogen	Polyethylene	Cool 4 °C	24 h
Organic particulates	Plastic Petri dish	Filter using GF/C filter,	6 months

		Cool 4 °C	
Organophosphorus pesticides	Glass	Cool, 4 °C, 10% HCl to pH 4.4	No holding, extraction on site
Pentachlorophenol	Glass	H_2SO_4 to pH < 4, 0.5 g $CuSO_4$ F[1] sample; Cool 4 °C	24 h
pH	Polyethylene	None	6h
Phenolics	Glass	H_3PO_4 to pH < 4, 1.0 g $CuSO_4$ r[1] sample; Cool 4 °C	24 h
Phenoxy acid herbicides	Glass	Cool 4 °C	Extract immediately
Phosphorus			
Dissolved	Glass	Filter on site using 0.45 μm filter	24 h
Inorganic	Glass	Cool 4 °C	24 h
Total	Glass	Cool 4 °C	1 month
Potassium	Polyethylene	Cool, 4 °C	7 days
Residue	Polyethylene	Cool, 4 °C	7 days
Selenium	Polyethylene	1.5 ml Conc. HNO_3 l^{-1} sample	6 months
Silica	Polyethylene	Cool, 4 °C	7 days
Sodium	Polyethylene	Cool, 4 °C	7 days
Electrical conductivity	Polyethylene	Cool, 4 °C	24 h
Sulphate	Polyethylene	Cool, 4 °C	7 days
Zinc	Polyethylene	2 ml Conc. HNO_3 l^{-1} sample	6 months

[1] Teflon containers can also be used to replace either the polyethylene or glass containers shown in the table.
Source: Adapted from Environment Canada, 1981

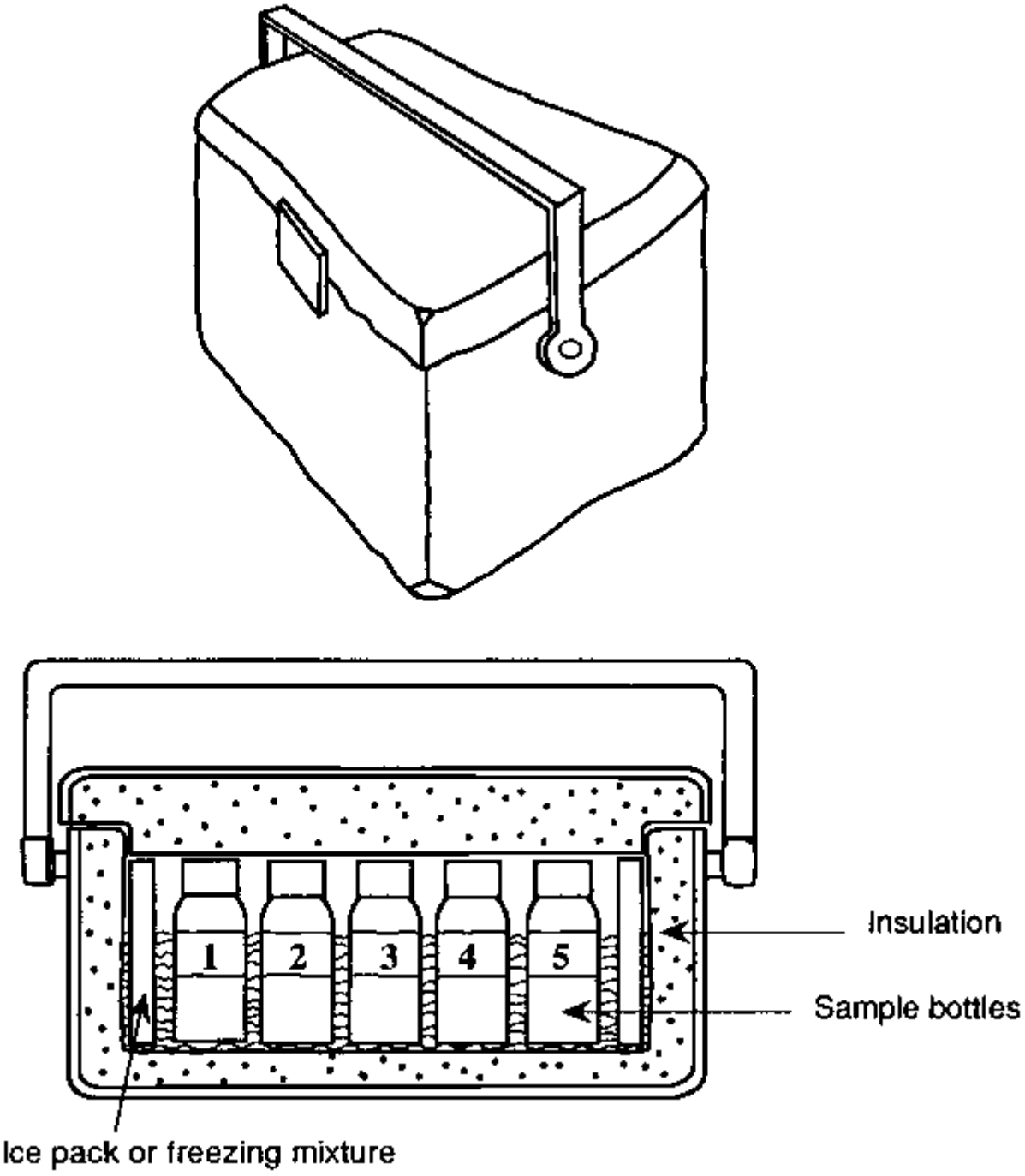
This information, as discussed earlier, will also be recorded in the field notebook.

Sample bottles should be placed in a box for transport to the laboratory. Sturdy, insulated wooden or plastic boxes will protect samples from sunlight, prevent the breakage of sample bottles, and should allow a temperature of 4 °C to be attained and maintained during transport. Figure 5.8 shows a suitable transport box. Rapid cooling of samples for BOD and/or microbiological analyses requires that the transport box should contain cold water in addition to ice or an “ice pack”. The use of a solid coolant alone is inadequate because heat transfer and sample cooling are too slow. Bottles containing samples for bacteriological analysis should ideally be placed in clear plastic bags to protect them from external contamination.

If the delay between sample collection and bacteriological analysis will be less than 2 hours, samples should simply be kept in a cool, dark place. When more than 2 hours will elapse, samples should be chilled rapidly to about 4 °C by placing them in a cold water/ice mixture (see above) in an insulated container, where they should remain during shipment. If the time between collection and analysis exceeds 6 hours, the report of the analysis should include information on the conditions and duration of sample transport.

In practice, it is difficult to ensure the transport of samples under conditions that do not affect their bacteriological quality and equipment designed for conducting analyses in the field is, therefore, becoming increasingly popular. It is also possible to filter samples in the field and to place the filters on a holding medium for later treatment in the laboratory.

Figure 5.8 Sample transport box



On arrival at the laboratory, samples for bacteriological analysis should be placed in a refrigerator and analysis should be started within 2 hours. Any samples arriving more than 24 hours after they were collected, or arriving unchilled more than 2 hours after they were collected, should be discarded. Analysis of such samples is unlikely to reflect the bacteriological condition of the water at the time of sampling.

Samples for chemical analysis should arrive at the analytical laboratory and be analysed within 24 hours of collection, since some variables are subject to change during storage (although others, such as hardness, fluoride, chloride and sulphate, are stable for 2 - 3 weeks).

5.8 Reception of samples by the laboratory

It is good practice for a member of the laboratory staff to sign for the receipt of samples and to make the following checks at the same time:

- All of the necessary details are recorded on the labels of sample bottles.

- The samples are contained in appropriate bottles.

- Samples have arrived in time for subsequent analysis to provide a reliable picture of water quality at the time of sampling.

- Samples have been treated with any necessary preservatives.

- Samples have been stored at appropriate temperatures, maintained throughout transport.

Samples should be logged into the laboratory system as soon as they arrive and (generally) transferred to a refrigerator at about 4 °C. If someone other than laboratory staff is to receive the samples, they should be instructed to transfer them directly to a refrigerator, noting the time and the condition of the samples, and then inform laboratory staff accordingly.

Where sample storage times and/or conditions have been such as to make it unlikely that analyses will yield reliable results, laboratory staff should decline to accept the affected samples. Supervisory staff should lend their support to the decision to reject samples on this basis.

5.9 Safety during field work

Field staff will encounter a wide range of hazards in the course of their work. To give just a few examples, water-courses may be highly contaminated with sewage or chemicals, access to sampling stations may involve crossing dangerous terrain, and wading in streams inevitably carries the possibility of slipping and personal injury. Where there is a risk of infection from contact with water (as in the case of schistosomiasis, for example), suitable protective clothing, such as rubber gloves, should be provided and its use by staff strongly encouraged.

Field staff should be trained to recognise and deal with as many as possible of the hazards they are likely to encounter. As a minimum, training should include water safety and first-aid. A basic first-aid kit should be carried at all times, and should not be left in the transport vehicle if staff are obliged to move any significant distance from it.

5.10 Source literature and further reading

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