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OXFAM DELAGUA

PORTABLE WATER TESTING KIT



USERS MANUAL

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OXFAM - DELAGUA

Portable Water Testing Kit

Users Manual

Revised and updated 3rd edition
1993

This equipment was designed to conform with the parameters specified in *World Health Organisation Guidelines for Drinking Water Quality Volume III*.

The equipment should only be used by trained personnel familiar with those guidelines.

For more details please contact WHO or The Robens Institute.

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OXFAM - DELAGUA Water Testing Kit - Users Manual

Copies of this manual can be obtained in the following languages:

English (3rd Edition)
Spanish (2nd Edition)
French (1st Edition)
Mandarin Chinese (2nd Edition)

If you regularly use the OXFAM - DELAGUA water testing kit and have translated the manual into another language, please send us the translation. Under these circumstances, we normally organise printing and give free copies to the programme which provided the translation.

For advice and information on the OXFAM - DELAGUA water testing kit, spare parts or service please contact us:

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We are continually trying to improve the OXFAM - DELAGUA water testing kit and because of this, some components may be different from those which appear in the manual.

The Robens Institute runs one and two week training courses either in the UK or overseas which include: water quality testing; sanitary inspection; water supply disinfection and use and maintenance of the OXFAM - DELAGUA water testing kit. Purchasers of the kit are also entitled to participate in a one day course at the Robens Institute in the use of the kit, free of charge.

The cover photograph of the kit being used in Mexico City on a hygiene education programme supported by the British Red Cross, is by Martin Fryer.

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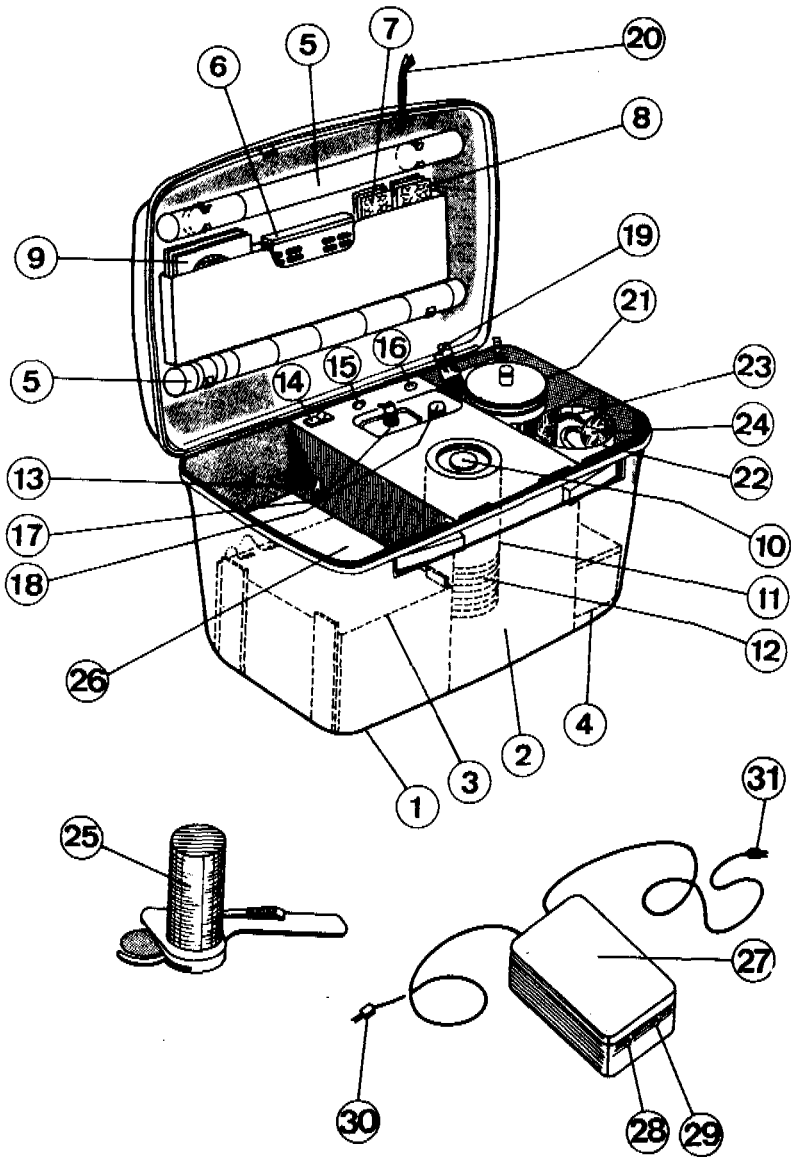
The OXFAM - DELAGUA Kit Components

Water Testing Kit

1. Case
2. Incubator
3. Battery
4. Spares Case
5. Turbidity Tubes (pair)
6. Chlorine and pH Comparator
7. Chlorine Test Tablets
8. pH Test Tablets
9. Membrane Filters
10. Incubator Lid
11. Incubator Pot
12. Petri Dishes
13. Power Socket
14. On/Off Switch
15. Power 'On' Indicator
16. Heater 'On' Indicator
17. Methanol Dispenser
18. Culture Medium Bottle
19. Lighter
20. Tweezers
21. Filtration Assembly with Sample Cup
22. Vacuum Cup
23. Sample Cable
24. Vacuum Pump
25. Absorbent Pad Dispenser
26. Storage Space

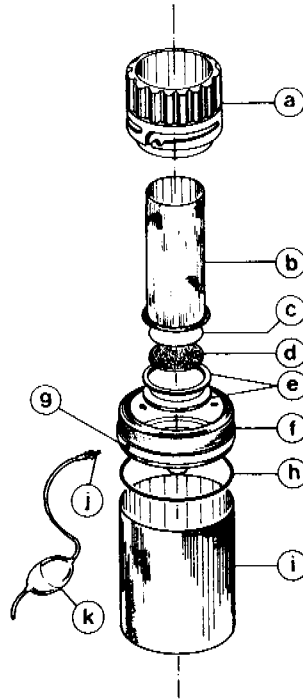
Charger Unit

27. Battery Charger/Mains Power Unit
28. Power 'On' Indicator
29. Charged battery Indicator
30. Incubator Plug
31. Mains Supply Plug



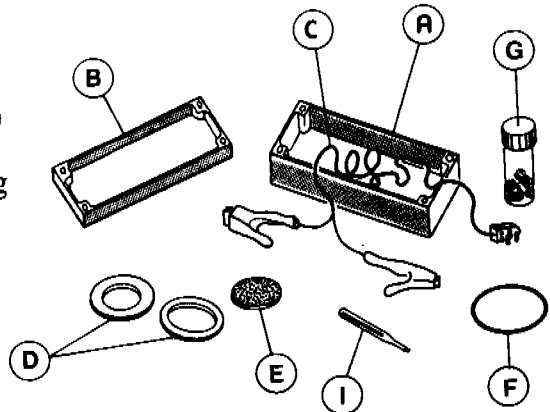
Filtration Apparatus and Components

- a. Plastic Collar
- b. Funnel
- c. Membrane Filter
- d. Bronze Disc
- e. Silicone Rings (pair)
- f. Aluminium Base
- g. Vacuum Connection
- h. Black Rubber O-Ring
- i. Vacuum Cup
- j. Vacuum Pump Connector
- k. Vacuum Pump



Contents of the Spares Case

- A. Base
- B. Lid
- C. External Battery
Connection Cable
- D. Silicone Rings (pair)
- E. Bronze Disc
- F. Black Rubber O-Ring
- G. Silicone Grease



Materials Needed for Testing

Before you start to use your OXFAM - DELAGUA water testing kit, you will need the following materials:

For preparation of culture medium:

- Pressure cooker, portable steriliser or autoclave
- Electric heating element, gas burner, stove or similar
- Distilled water (alternatives are discussed in the text)
- Means of measuring distilled water eg. measuring cylinder

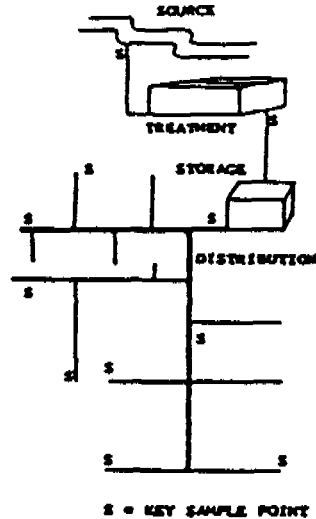
For using the kit in the field:

- Methanol (alternatives are discussed in the text)
- Paper towels or clean cloths
- Wax pencil or marker pen
- Report sheets (see page 56 for an example)

Sampling Programmes

Selection of Sites and Frequency of Sampling

Samples should be taken from locations which are representative of the water distribution network and household connections. Where there are several sources and a mixed distribution system, it is necessary to take account of this. Where there is a branched distribution system, samples should be taken at random points evenly spread throughout the system. Where there are main branches and a remote periphery (as shown) greater attention should be devoted to the main branches and remote points in the network.



The recommended minimum frequencies for sampling both for piped supplies and point sources are shown in the tables below.

Minimum Frequency of Sampling and Analysis of Piped Water Supplies

| Population served | Minimum frequency of sampling |
|-------------------|--|
| Less than 5,000 | 1 sample monthly |
| 5,000 to 100,000 | 1 sample per 5,000 population monthly |
| more than 100,000 | 20 samples monthly plus 1 sample per 10,000 population monthly |

Minimum frequency of Sampling and Analysis of Unpipied Water Supplies

| Source and mode of supply | Bacteriological | Physical/ Chemical | Remarks |
|---|--|---|---|
| Open well | Nil | Once initially for community wells | Pollution usually expected to occur |
| Covered well shallow tubewell with handpump | Nil | Once initially thereafter as situation demands | Testing needed when environmental conditions change or when outbreak or increase in waterborne disease occurs |
| Deep tubewell with handpump | Once initially thereafter as situation demands | Once initially thereafter as situation demands | Testing needed when environmental conditions change or when outbreak or increase in waterborne disease occurs |
| Springs and piped supplies | Once initially thereafter as situation demands | Test periodically for residual chlorine if water is chlorinated | Testing needed when environmental conditions change or when outbreak or increase in waterborne disease occurs |
| Community rainwater collection systems | Sanitary protection measures and bacteriological testing only if situation demands | Not needed | |

Source: Adapted from *WHO Guidelines for Drinking Water Quality Volume III* Geneva 1985.

Preparation of the Kit

Preparation of Culture Medium in the Laboratory

You will need the following items:

- Membrane Lauryl Sulphate Broth
- Distilled water
- Polypropylene bottles
- Measuring cylinder
- Pressure cooker, steriliser or autoclave*
- Heating element, stove or burner

*Note: A portable steriliser kit is available from the Robens Institute

Carefully wash the plastic culture medium bottles in clean, water before use. If necessary, use a little detergent and then rinse well with clean water to remove all traces of the detergent.

1. Place 38.1g of Membrane Lauryl Sulphate Broth powder into a beaker and add 500ml of distilled water or clean, filtered rainwater. The broth powder is usually supplied in pre-weighed tubs of 38.1g. Tubs of 500g are available for those needing large quantities of culture medium. It is important to keep the powdered medium well sealed in its container until used as the powder absorbs water and will deteriorate if left open to the atmosphere.
2. Mix and apply gentle heat if necessary to dissolve the powder completely. *Do not boil*. The culture medium will be a bright red colour when dissolved.
3. Pour suitable volumes of culture medium into the polypropylene bottles. You will need approximately 2.5ml of medium per sample. Each bottle should contain enough medium for one days use. The kit contains enough dishes to process 16 samples per day.
4. Replace the screw caps and tighten carefully. Over-tightening can result in leakage.

5. If an autoclave is available, sterilise the bottles with the tops loose at 121°C for 10 minutes.

If there is no autoclave available, then a household pressure cooker or portable steriliser may be used. Place the bottles in a rack (they may melt if placed directly on the floor of the cooker) replace the lid and heat to full pressure (about 15psi). Once the cooker has reached full pressure, time 15 minutes using a stopwatch or clock. Switch off the heat and allow the cooker to cool. Remove the bottles and store in a cool, dark place.

Preparation of Culture Medium in the Field

You will need the following items:

Membrane Lauryl Sulphate Broth
Clean water
Polypropylene bottles
Cooking pot or pan

Carefully wash the plastic culture medium bottles in clean, warm water before use. If necessary, use a little detergent and then rinse with clean water to remove all traces of the detergent.

1. Use distilled water if possible. If this is not available, obtain the cleanest water possible eg. rain water or filtered and boiled water. If necessary, stand the water in a container overnight to settle out any particles. *Never use chlorinated water.*
2. Using the pH tester in the kit, check that the pH of the water is between 6.8 and 8.2. If it is not, it will be necessary to find an alternative source.
3. Measure out 500ml of clean water in a beaker.
4. Add 38.1g of Membrane Lauryl Sulphate Broth to the 500ml of water in the beaker. Mix and heat gently if necessary to dissolve the powder. *Do not boil*

5. Pour suitable volumes of culture medium into the polypropylene bottles. You will need approximately 2.5ml of medium per sample. Each bottle should contain enough medium for one days use. The kit contains enough dishes to process 16 samples per day.
6. Replace the screw caps and tighten carefully. Over-tightening can result in leakage.
7. If a pressure cooker is available, sterilise the culture medium as described in 5. at the top of page 10.
8. If no pressure cooker or portable steriliser is available, place the bottles of culture medium into a cooking pot or pan of boiling water taking care to ensure that the bottles do not come into contact with the base of the pan (use a rack or stand). Boil for 20 minutes. Store the sterilised bottles in a cool, dark place. Use any culture medium prepared in this way within 24 hours.

Storage of Culture Medium

Culture medium which has been sterilised in an autoclave or pressure cooker and then stored in a cool dark place normally remains stable for several months. If signs of deterioration appear eg. cloudiness or yellow colouration, the contents of the bottle must be discarded. In cases where the medium is kept in a cold environment eg. refrigerator, a deposit may form which dissolves when the bottle is warmed and gently shaken. This deposit does not mean that the medium has deteriorated, it is a precipitate of lauryl sulphate.

Disposal of Contaminated Material

Contaminated material such as used membranes and pads, may be made safe by autoclaving or incineration. The petri dishes must be carefully washed after use with detergent followed by rinsing with clean water and drying.

Sterilisation of the dishes may be carried out in several ways:

1. Autoclave at 121°C for 10 minutes. Assemble the dishes and store away from sources of contamination.
2. Place the dishes in a conventional oven at 180°C for 30 minutes.
3. Plunge the bases and lids of the dishes into boiling water for 15 minutes. Pour away the water and assemble the dishes as they dry, while they are still hot.

Whenever it is possible, always use one of the above methods. If this is not possible then the following method can be applied:

1. Flame the bases and lids of the dishes with the lighter or gas burner using the tweezers to hold the dishes. Assemble while still hot.

Absorbent Pads and Dispenser

The pads are supplied sterile in packs of 100 units. A sterile pad dispenser is also supplied with the kit. Never leave the dispenser without a pack of pads attached as it will become contaminated.

It is preferable to dispense pads into the petri dishes at base and not to use the dispenser and pads in the field.

If it is necessary to dispense pads in the field, take care not to contaminate the dispenser assembly. If the dispenser is lost or damaged, pads may be dispensed in the field using the sterile tweezers. Some kit operators prefer this method instead of using the dispenser.

Methanol Dispenser

The methanol dispenser is supplied with a plastic cap and dispensing nozzle. The dispenser should be half-filled with methanol using a small funnel, pipette or syringe to avoid spillage of methanol. *Do not overfill the methanol dispenser as it may leak in hot weather.*

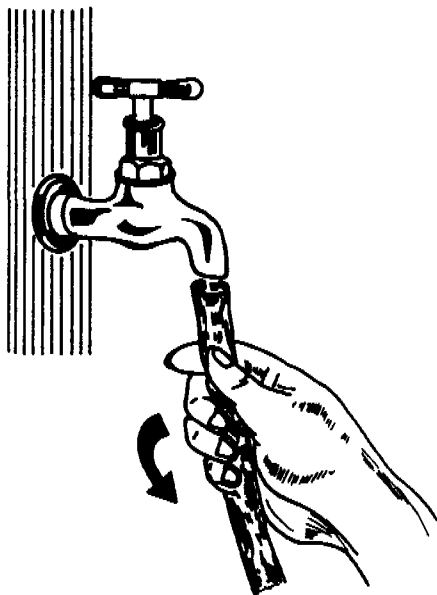
The plastic cap is a push-fit onto the methanol dispenser. However, the cap may be very tight when new and be difficult to fit. To make fitting and removal easier, it is helpful to submerge the cap in hot water for a few minutes to soften the plastic, it will then be easier to fit and remove.

To dispense methanol, lever the dispensing nozzle into the upright position with the tip of the tweezers. To seal off the flow of methanol, push the nozzle down into the recess in the cap. Be sure not to leave the dispensing nozzle in the upright position after using the kit as the methanol will leak.

Sampling Methods

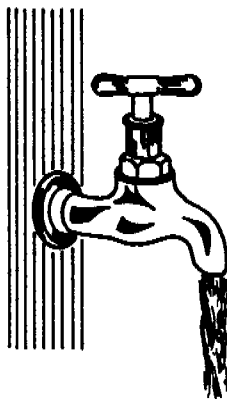
Sampling from a Tap

1. Remove any attachments eg. nozzles, pipes etc. from the tap. Check that the tap does not leak and that all seals are in good condition.

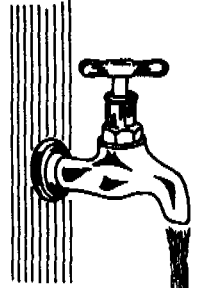


2. Carefully clean the mouth of the tap with a clean cloth or tissue to remove any dirt or grease.

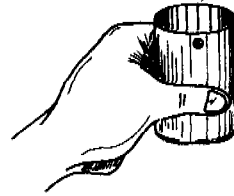
Open the tap and leave running for at least one minute before taking a sample. This ensures that any deposits in the pipes are washed out.



3. Take a water sample with the non-sterile vacuum cup. Analyse the sample for chlorine residual and turbidity levels.

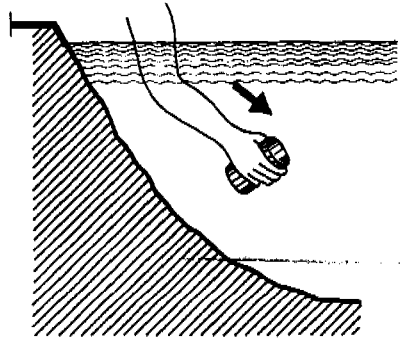


4. If the chlorine residual and turbidity results suggest, then take a second sample for bacteriological analysis using the sterile sample cup.

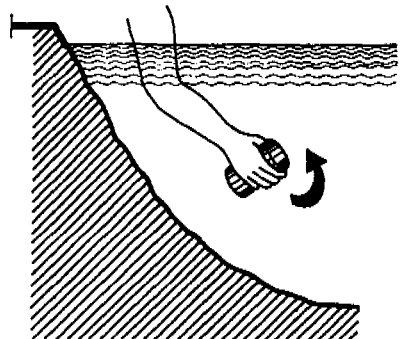


Sampling from a Lake, Reservoir or other Surface Water Source.

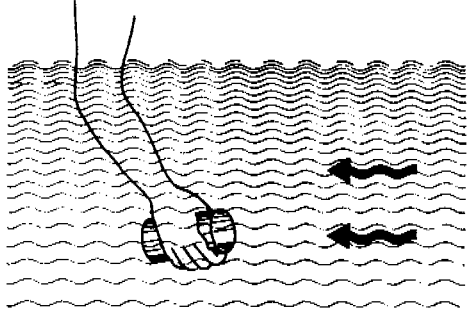
Where there is adequate access it may be possible to take samples by hand. In many cases it may be inconvenient or dangerous to enter the water. Grasp the sample cup firmly and dip the open mouth of the cup into the water. Submerge the cup to about 30cm below the surface of the water and scoop up the water sample.



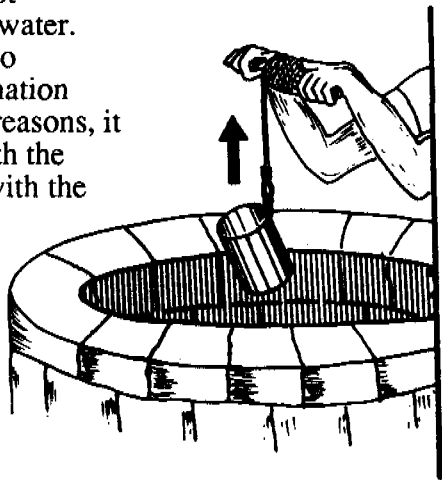
This scooping action ensures that no external contamination enters the sample cup.



In areas where there is a current flow eg. rivers and streams, the sample should be taken against the current flow.

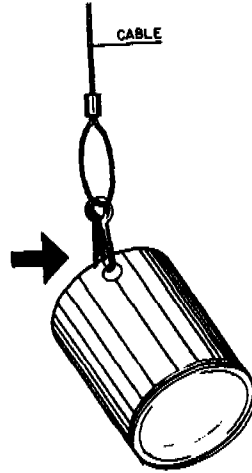


In all cases, it is vital to obtain a sample which is representative of the main body of water. For example, when sampling from a river, do not sample the quiet or stagnant areas near the bank as these do not represent the main body of water. Furthermore, it is vital not to introduce external contamination into the sample. For these reasons, it is often better to sample with the help of the cable supplied with the kit.

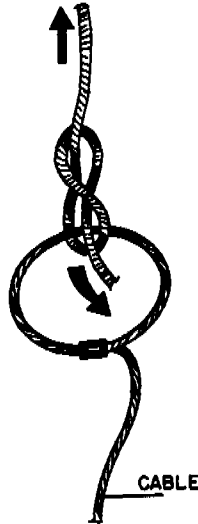


Sampling from an Open Well or Storage Tank

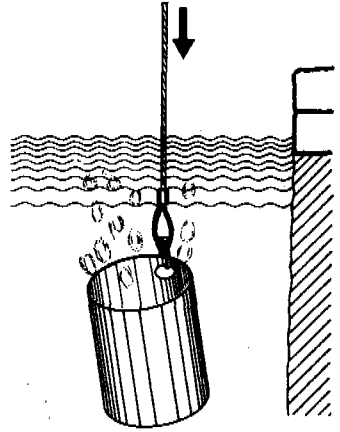
1. Fasten the sampling cable to the hole in the lip of the sample cup by means of the clip on the end of the cable.



It may be necessary to increase the length of the cable by attaching a rope or string to the sample cable. Take care not to lose the sample cup.



2. Lower the sterile sample cup into the well or tank, taking care not to allow the cup to touch the walls of the structure where it may pick up dirt. Submerge the cup to a depth of 30cm.



3. Lift the sample cup carefully and place on a clean surface where it cannot be knocked over.

Processing of Samples Using the Kit

Introduction

The first tests which must be carried out on a drinking water sample are the determination of chlorine residual and turbidity. The sample must be taken in a clean, but non-sterile cup eg. the vacuum flask, which has been rinsed several times before taking the sample for analysis.

If the results of the analysis are as follows:

Residual free chlorine greater than 0.2mg/litre (0.2ppm) *and*

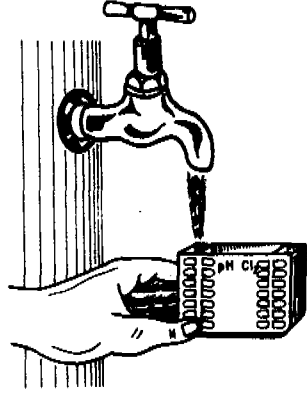
Turbidity less than 5TU

It is highly *unlikely* that the sample will contain thermotolerant (faecal) coliform bacteria and therefore it may not be necessary to carry out thermotolerant (faecal) coliform analysis.

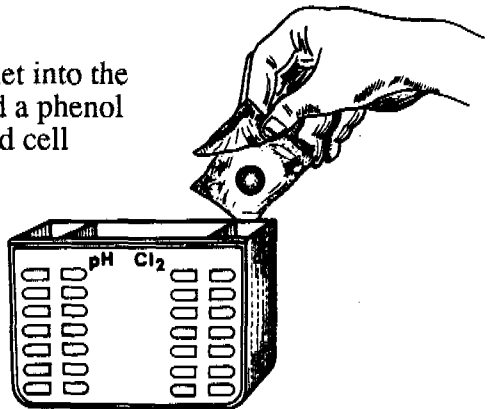
Where results are not within the above criteria, it is necessary to carry out thermotolerant (faecal) coliform analysis. Under these circumstances, samples for analysis must be taken with the sterile sample cup.

Analysis of Chlorine Residual and pH

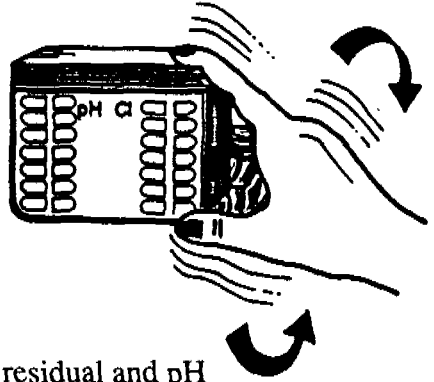
1. Wash the comparator cells three times with the water which is to be analysed and finally fill all three cells with the sample.



2. Drop a DPD No. 1 tablet into the right-hand cell ('Cl₂') and a phenol red tablet into the left-hand cell ('pH').



3. Replace the lid of the comparator and push down firmly to seal. Invert the comparator repeatedly until the two tablets have dissolved completely.



4. Immediately read the free chlorine residual and pH concentrations by holding the comparator up to daylight and matching the colour developed in the cells with the standard colour scale in the central part of the comparator. If the colour falls between two standard colours then it will be necessary to estimate the concentration. Record the result on the daily report sheet (See page 56 for an example).

5. To test for total chlorine residual, do not discard the liquid in the comparator, but remove the lid and add a tablet of DPD No.3 to the right hand cell ('Cl₂').

6. Invert the comparator repeatedly to dissolve the tablet. The colour developed after 10 minutes represents total chlorine residual in mg/litre.

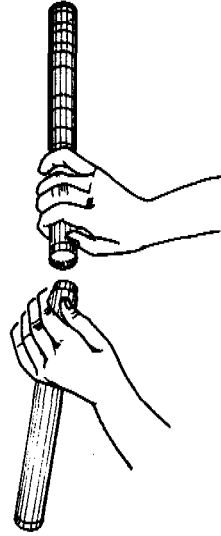
7. Subtract the free chlorine result from the total chlorine result to obtain the combined chlorine concentration:

| | | |
|-----------------------|---|-------------------------|
| DPD No. 1 | = | Free chlorine residual |
| DPD No. 1 + DPD No. 3 | = | Total chlorine residual |
| Total - Free | = | Combined |

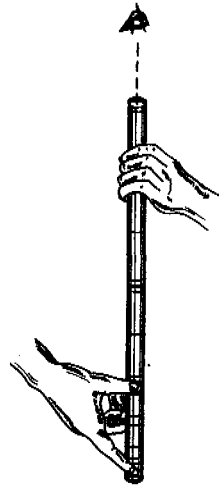
Turbidity Analysis

Note: These turbidity tubes cover the range 5 to 2,000 TU

1. Remove the two turbidity tubes from their clips in the lid of the case. Carefully push the upper tube (open at both ends) squarely into the lower tube and look through the open end of the tube at the black circle printed on the base of the tube. Ensure that there is good illumination available. Normal daylight is adequate for this purpose.



2. Pour the water sample into the tube from the sample cup until the black circle just disappears when viewed from the top of the tube. Avoid creating bubbles as these may cause false readings. Do not strain to see the black circle as this can sometimes cause biased results.



3. The turbidity tubes are graduated in a logarithmic scale with the most critical values. The result is the value of the line nearest to the water level. This permits a reasonably exact estimation of the turbidity of the water sample.



Bacteriological Analysis of Water

The analysis of water samples for thermotolerant (faecal) coliforms is carried out by passing a measured quantity of water through a sterile filter. Any bacteria present in the water are caught in the filter. The filter is then placed onto a paper pad soaked in a liquid growth medium which feeds coliform bacteria, but inhibits any other bacteria caught in the filter. To ensure that only thermotolerant (faecal) coliform bacteria are allowed to grow, the filter is kept at 44°C in the kits' incubator until the bacteria multiply many times to form colonies of bacteria which can be seen with the naked eye. Thermotolerant (faecal) coliforms are recognised by their ability to produce a colour change (from red to yellow) in the culture medium at 44°C. Results are expressed as colony-forming units per 100ml of water (CFU/100ml).

Thermotolerant (faecal) coliforms are of sanitary significance when present in drinking water supplies. Users should refer to country specific water quality standards or guidelines or to World Health Organisation *Guidelines for Drinking Water Quality Volume III* to decide when action should be taken to improve contaminated water supplies.

Some users may need to analyse for total coliform bacteria which, although of less sanitary significance than thermotolerant (faecal) coliforms, can be used to indicate problems in large distribution networks. Total coliform analysis is carried out using the same process as for thermotolerant (faecal) coliforms, the only difference being that the filters are incubated at 37°C.

The OXFAM - DELAGUA incubator can be recalibrated to 37°C following the recalibration procedure on pages 52 to 54. However, this is not satisfactory when carrying out both thermotolerant (faecal) coliform and total coliform analysis on a regular basis. A dual temperature incubator is available from the Robens Institute which allows both tests to be carried out simultaneously .

Selection of Appropriate Sample Volumes for Thermotolerant (Faecal) Coliform Analysis

The most appropriate volume to process is that which allows the most accurate count of the bacteria. The most statistically valid range of colonies to count for contaminated water supplies is between 20 and 200 colonies per membrane filter. When there are less than 20 colonies there is a possibility of statistical error and it may prove difficult to count more than 200.

Water for Human Consumption

For treated waters or water in a distribution network it is likely that the number of thermotolerant (faecal) coliforms per 100ml will be around zero. For these waters the standard volume used is 100ml and a count of zero thermotolerant (faecal) coliforms per 100ml is indicative of a safe water supply. If the count exceeds 50 colonies per 100ml then the supply is heavily contaminated and requires immediate remedial action. The same action is required when waters treated with disinfectant eg. chlorine, give a count of greater than 1 thermotolerant (faecal) coliform per 100ml.

Other Water Sources

The selection of the most appropriate sample volume for a given source, treatment plant or distribution system is normally best made in the light of previous experience.

For piped supplies and partially treated waters (including those derived from ground water sources), it is possible to adjust the volume of sample to obtain a final count in the range 20 to 200 faecal coliforms per 100ml. Recommended volumes for each type of source are shown below:

Suggested volumes for thermotolerant (faecal) coliform analysis by the membrane filtration technique (alternative volumes are shown in brackets).

| | |
|---|-------------------|
| Lakes, ponds and other surface waters | 10(1ml*) |
| Protected ground waters eg. wells and springs | 100(50ml) |
| Unprotected ground waters eg. open dug wells and springs | 50(10ml) |
| Waters in treatment plants after partial treatment | 50(100ml or 10ml) |
| Waters in treatment plants after full treatment | 100ml |
| Reservoirs, distribution networks and household taps | 100ml |

*Note: This volume will require the use of sterile pipettes and dilution water.

These are guide values; they do not represent absolute recommendations to be applied to sampling programmes. It may be useful to analyse different volumes of the same sample in order to decide the best range in which to count the bacteria. It is not necessary to sterilise the filtration apparatus and sample cup between two analyses of the same sample provided that the smaller volume is processed first.

General Hygiene in the Field

Although all components of the kit should be kept free from dirt and other contamination, there are some parts of the kit which must always be kept *clean and sterile*. These are as follows:

a) All those areas in direct contact with the water sample, eg. the internal surface of the sample cup, the internal surface of the filter funnel, the upper part of the filtration base and the surface of the bronze disc.

b) Surfaces in contact with the culture medium, eg. the internal surface of the petri dishes and the absorbent pads.

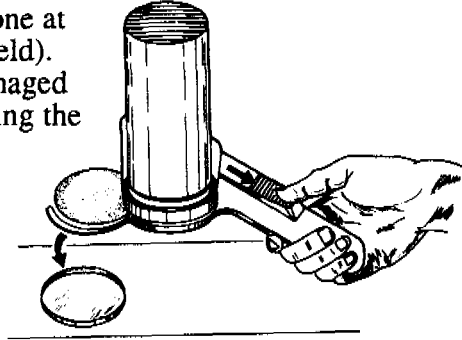
c) Parts in contact with the membrane filters, eg. the filtration apparatus, the absorbent pads and the tweezers.

Under no circumstances should any of these components be allowed to come into contact with dirt, dust or external objects which may contaminate them and interfere with the bacterial count.

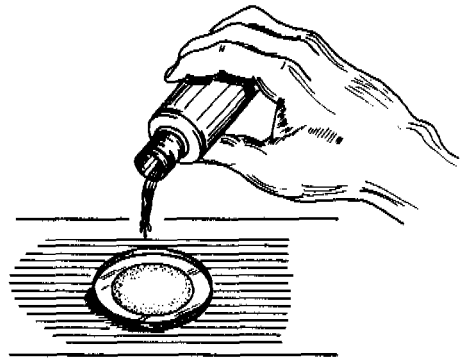
Before handling a membrane filter and after processing a sample, the tips of the tweezers should be flamed with the lighter. Hold the tips in the flame for five seconds and allow to cool before handling a membrane filter. After sterilising the tweezers in this way, they should be placed so that the tips do not touch any other object.

Sample Processing for Thermotolerant (Faecal) Coliform Analysis in the Field

1. Using the absorbent pad dispenser, place one pad into each petri dish (this is normally done at base before leaving for the field). If the dispenser becomes damaged the pads can be dispensed using the tweezers.

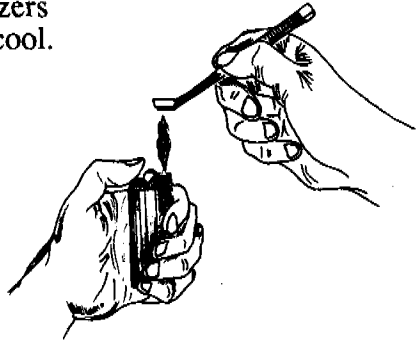


2. Pour enough culture medium to soak the pad and leave a slight excess, onto the absorbent pad in the petri dish (approx 2.5ml). Replace the bottle cap immediately. Do not allow the bottle neck to come into contact with any external objects. Immediately before processing a sample, drain off most of the excess medium by inverting each dish with the lid removed. Always ensure that a slight excess remains to prevent drying of the pad during incubation.

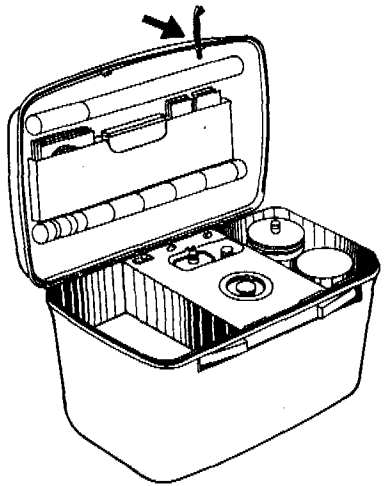


Note: Once the bottle of culture medium has been opened it is recommended that the contents are used within one day. It is not advisable to use the medium in one bottle over several days since this can lead to contamination.

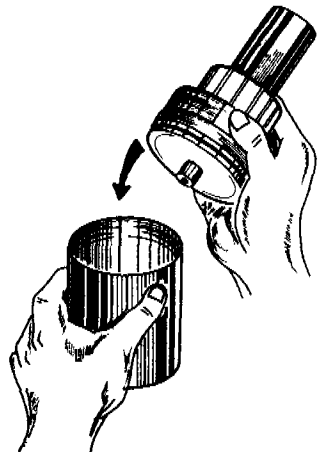
3. Flame the tips of the tweezers with the lighter and leave to cool.



4. Place the heel of the tweezers into the test kit case as indicated. This ensures that the tips are kept away from all sources of contamination whilst analyses are in progress.

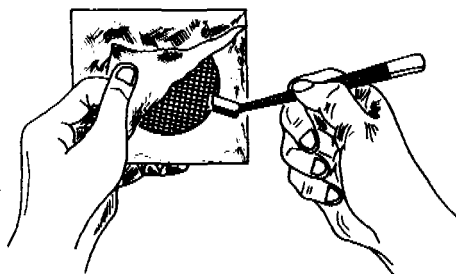


5. Remove the sterile filtration cup from the filtration apparatus. Push the filtration apparatus firmly onto the vacuum cup (if this is difficult, then it is likely that the sealing ring needs lubrication with silicone grease - see page 42 for maintenance of the kit). Place the assembly in an upright position in a convenient place in the kit. Do not place the apparatus on the ground where it may become soiled.

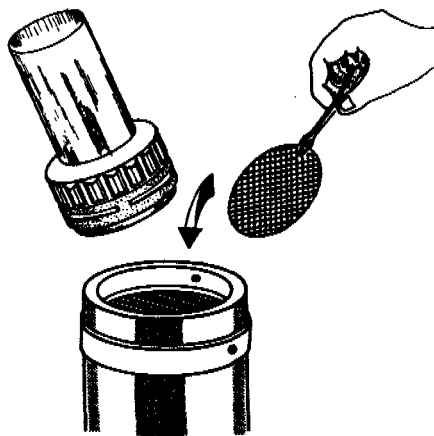


6. Unscrew the plastic collar and filtration funnel in order that these may be easily removed. Do not place these on any surface other than the filtration base.

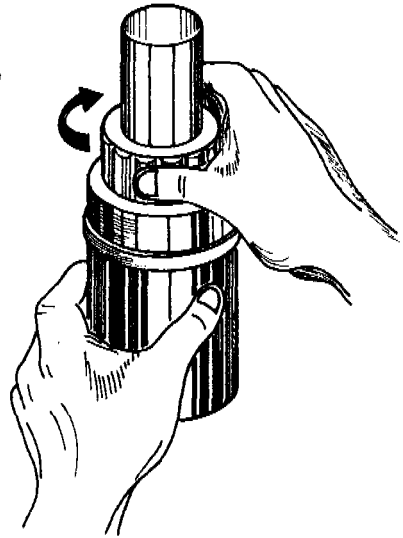
7. Using the sterile tweezers, carefully remove a sterile membrane filter from its packet. Hold the membrane only by the edge.



8. With one hand lift the filtration funnel and plastic collar above the filtration base. With the tweezers in your other hand, place the membrane filter (grid side uppermost) onto the bronze disc filter support. Replace the funnel and collar immediately without allowing them to come into contact with any external objects. It is normally convenient to hold the funnel between the thumb and forefinger which ensures that the collar will not slip off and that the fingers do not come into contact with the interior surface of the funnel.

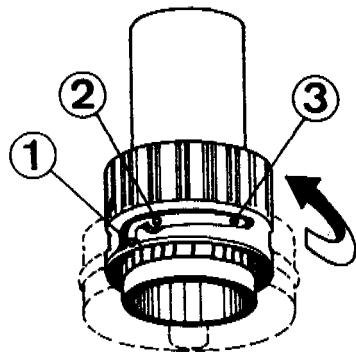


9. Screw the plastic collar down tightly to hold the membrane and to provide a water tight seal.



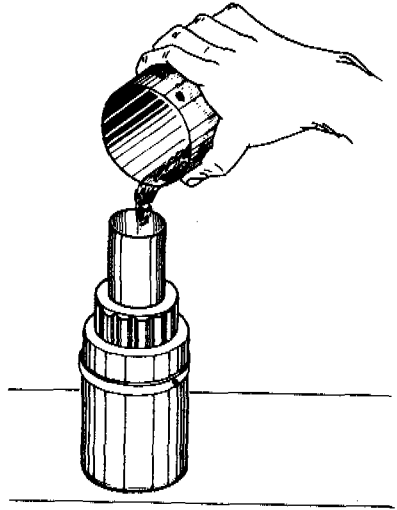
Note: The plastic collar has three adjustment positions:

1. Completely free - The apparatus can be dismantled when in this position.
2. Loose but not free - all interior surfaces are exposed to the atmosphere. This is the position used when sterilising the apparatus.
3. Fully tightened - the funnel forms a tight hermetic seal between the membrane support and the membrane filter. This is the position for filtration.

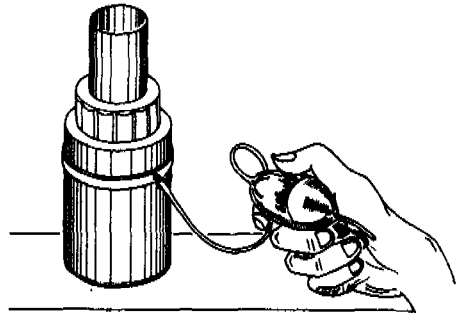


10. Rinse the sterile sample cup once with the water to be sampled and then fill the cup with the water. Take care not to allow external contamination to enter the sample cup eg. dirt and debris.

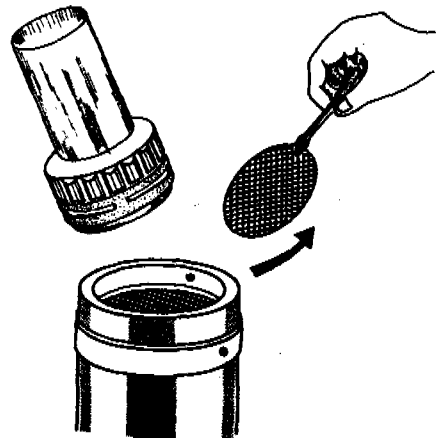
11. Pour the sample into the filtration funnel up to the appropriate mark (10, 50 or 100ml) engraved on the internal surface of the funnel. Take care not to allow external debris to enter the funnel.



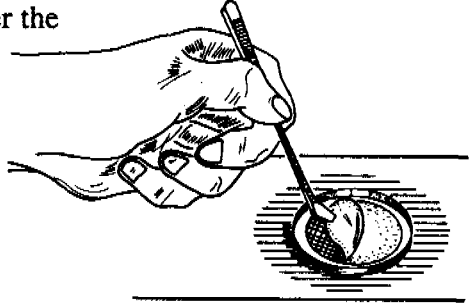
12. Insert the plastic connector of the vacuum pump into the vacuum connection on the filtration base. Squeeze the pump bulb several times to draw a vacuum and squeeze as required to draw all the water through the membrane filter. When all the water has passed through the filter, disconnect the pump from the filtration apparatus.



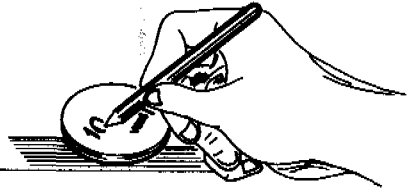
13. Unscrew the collar and remove the funnel and collar with one hand. Using the tweezers in the other hand lift the membrane carefully from the filtration base. Hold the membrane by the edge only.



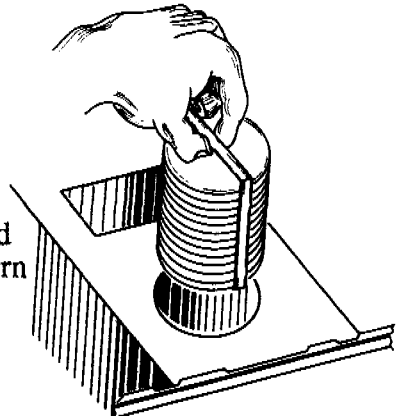
14. Remove the lid of a prepared petri dish and place the membrane (grid side uppermost) onto the absorbent pad soaked in culture medium. Start at one edge and lower the membrane by 'rolling' so as to avoid trapping air bubbles under the membrane.



15. Replace the lid of the petri dish and mark the lid with sample information eg. volume filtered, source, time and date or a code which relates to details on the daily report sheet. A wax pencil or marker pen is suitable for this purpose.



16. Place the petri dish with the lid uppermost into the carrier and return the carrier to the incubator pot. Replace the incubator lid.



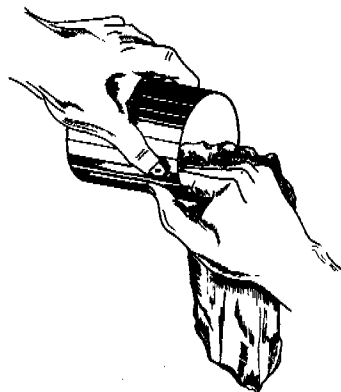
Re-Sterilisation of the Filtration Apparatus

The sample cup and the filtration apparatus must be re-sterilised between samples when analysing water from two different sources.

Sterilising the equipment in the field presents some practical problems and must be carried out using simple methods. The most appropriate is the use of *methanol* which is described below. Where methanol is not available, the filtration apparatus and sample cup can be sterilised by immersion in boiling water for five minutes.

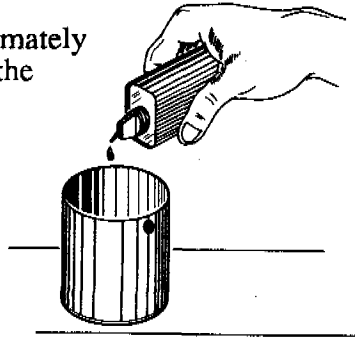
Note: Methanol is the *only* alcohol suitable for sterilising the filtration apparatus, there is no substitute. When burnt in a shortage of oxygen - in the closed sampling cup for example - formaldehyde gas is produced which is a very effective disinfectant. Methanol is expensive to freight and requires special transport conditions. It is therefore usually obtained in-country from a pharmaceutical supplier and is not normally supplied with the kit. However, methanol can be supplied for freighting by the Robens Institute on request.

1. Carefully dry the sample cup and the filtration assembly with a clean, dry towel or tissue.

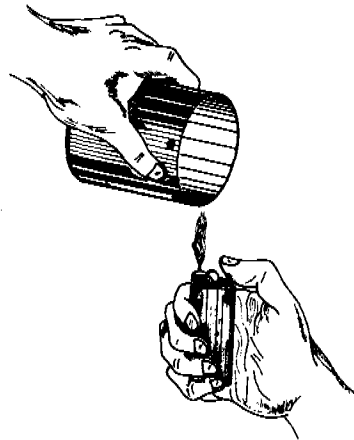


2. Using the plastic collar, secure the filtration funnel in the second position (see page 30) which allows the sterilising agent to penetrate.

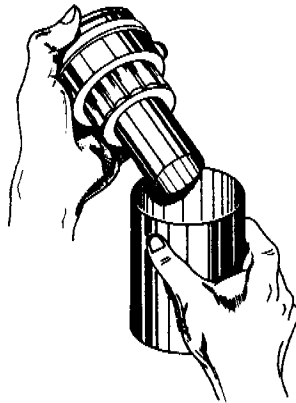
3. Pour about 1ml (approximately 20 drops) of methanol into the sample cup.



4. Carefully ignite the methanol in the sample cup using the lighter.
Caution: keep the mouth of the cup away from your face.
Place the cup on a flat surface which will not be harmed by heat.



5. Allow the methanol to burn for several seconds and when almost completely burned up, place the filtration apparatus over the sample cup and push firmly into place to form a good seal.



6. Keep the filtration apparatus in the sample cup for at least 15 minutes before using to process a further sample.

Note: The use of too much methanol will result in a residue being left in the sample cup and filtration apparatus after sterilisation. The ideal volume of methanol to use will be determined in the light of experience.

Note: It is best to sterilise the filtration apparatus immediately after each analysis and to keep the filtration apparatus in a sterile condition during transport and storage. In this way, the filtration apparatus is always ready for use.

Resuscitation of Bacteria

Once the last sample of the day has been taken, wait for a minimum of 60 minutes before switching on the incubator (resuscitation time). Try to plan the day so that the time between the processing of the first and last samples is no more than 3 hours. This restricts the resuscitation time to a maximum of 4 hours.

Note: Resuscitation time is particularly important for chlorinated waters or marine water where the thermotolerant (faecal) coliform bacteria are 'stressed' due to environmental exposure. For these types of waters it is beneficial to leave processed membranes for 4 hours after the last sample has been processed before switching on the incubator.

Sample Incubation

Incubate the samples for 16 to 18 hours. The incubator is designed to maintain a temperature of $44^{\circ}\text{C} \pm 0.5^{\circ}\text{C}$. In order to maximise on battery life, do not leave the incubator on for more than the specified period eg. 4.00pm to 8.00am.

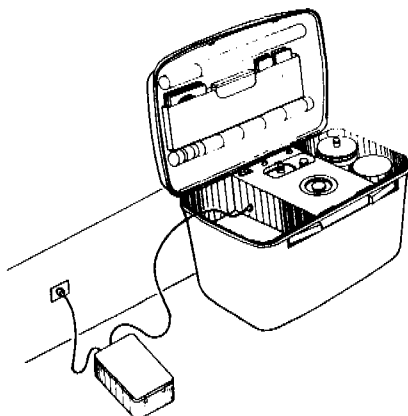
There are three alternative power sources for the incubator:

1. Mains electricity supply via the charger unit.
2. Internal battery.
3. External 12v battery.

Note: It is recommended that the mains supply option be used wherever possible. When used in this way, the charger unit will operate the incubator and charge the battery. If the mains electricity fails, the internal battery will operate the incubator automatically.

Using Mains Electricity or Generator via the Charger Unit

When using mains electricity to power the incubator, the incubator can be operated and the internal battery charged simultaneously. If the power fails for any reason, the internal battery continues the incubation cycle. When operating from mains electricity, connect the three pin plug to the socket in the left hand side of the incubator console. Plug the incubator into the mains electricity socket using an appropriate plug and switch on the mains. Switch on the incubator and leave until the incubation cycle is complete.



Internal Battery

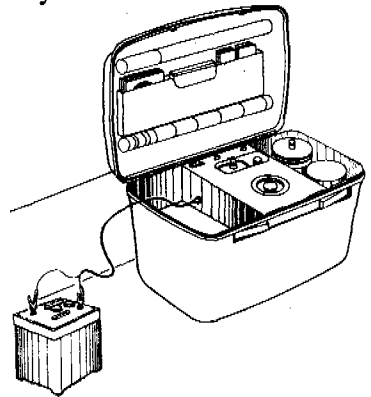
When planning to work for several days in the field, it is possible to obtain up to five incubation cycles from the internal battery. When using the internal battery in this way, *never* try to use the incubator for *more than five cycles* without recharging the battery or run the incubator for more than 18 hours during any cycle. Always recharge the battery fully at every opportunity using mains electricity.

External 12v Battery

When planning to work in the field for more than five days, or when working in remote areas it is possible to operate the incubator using an external 12v battery eg. vehicle battery using the connection lead provided in the spares case or the auxiliary battery pack available as an optional extra.

An external battery cannot be used to recharge the internal battery, only to operate the incubator. When incubating, very little current is drawn and it is usually safe to operate from a vehicle battery for one incubation cycle without risk of discharging the vehicle battery excessively.

To operate the incubator from an external battery, connect the crocodile clips on the battery lead to the correct terminals on the external battery (Red to Positive or '+' and Black to Negative or '-'). Connect the three pin plug to the left hand side of the incubator console. Switch on the incubator and check that the 'Power on' indicator is lit.



Note: A poorly maintained external battery may cause the internal battery to discharge.

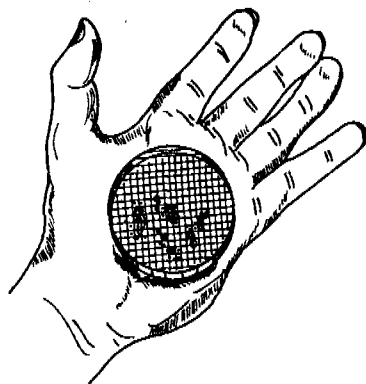
Note: Never run the incubator from a vehicle battery for more than one cycle if the vehicle is not being driven regularly. Repeated use of the incubator will drain a vehicle battery.

Always incubate the petri dishes with the incubator and case lids firmly closed. This reduces heat loss and saves battery power.

Always keep the kit in a normal environment eg. place on a chair or table to prevent heat loss through the floor and avoid incubating samples outdoors during cold weather.

Counting Colonies and Recording Results

1. Once the incubation period is complete, remove the petri dishes and their holder from the incubator pot. Remove the lid of a petri dish and observe the surface of the membrane in good incident light.
2. Count all the yellow colonies which have a diameter of between 1 and 3mm. Do not count colonies which are transparent or red/pink, on cooling. These are bacteria which do not ferment lactose and cannot be identified without further study. They are not thermotolerant (faecal) coliforms.



Colonies may vary considerably in size. Generally where the membrane contains a large number of colonies, the colonies are smaller in diameter. Where colonies are fewer, they tend to be larger. This is because the colonies compete for nutrients and will grow larger where there is less competition.

If there are large numbers of yellow colonies, count methodically using the horizontal grid lines. In this way it is possible to count between 1 and 200 colonies per membrane.

3. Convert the count into number of Thermotolerant (faecal) coliforms per 100ml and record the result on the daily report sheet.

The calculation is made as follows:

| Volume filtered | Thermotolerant (faecal) coliforms per 100ml |
|------------------------|--|
| 100ml | Number of colonies X 1 |
| 50ml | Number of colonies X 2 |

Care and Maintenance of the Kit

The Battery

Never

Allow the internal battery to discharge completely.

The useful life of the battery will be maximised if the battery is always kept in a well-charged state. In order to ensure this, it is advisable to recharge the battery fully every weekend wherever possible.

For this reason:

Never

Perform more than five incubation cycles without recharging the battery.

Never

Leave the incubator switched on for more than 18 hours.

Always

Incubate samples with the incubator lid firmly in place and the kit closed.

Always

Operate the incubator indoors or in a vehicle and on a chair or table to prevent heat loss through a cold floor. Do not operate outside in cold weather conditions.

Always

Recharge the internal battery at the end of a period of work in the field.

Always

Leave the battery in a charged state when the kit is out of use or in storage. During storage, recharge monthly.

To recharge the battery, connect the small three pin plug from the charger to the left hand side of the incubator console. Plug the charger into the mains electricity supply and switch on. Check that the incubator is switched off, unless it is in use. Leave the kit and charger until the green light on the charger goes off indicating that the battery is completely charged. This process may take 12 to 36 hours depending on the state of charge of the battery. When the battery is completely charged, switch off the charger, disconnect from the mains electricity supply and the incubator and store in a safe place.

When using the kit in low temperature environments eg. less than 10°C, the maximum number of incubation cycles on one battery charge should not exceed three to four.

If any faults or malfunctions are apparent in the kit, refer to the section on 'Fault Finding in the Incubator, Battery and Charger' (See Page 43).

Electronic Components and the Incubator

Do not allow water to enter the base of the kit

The electronic components are sealed and tropicalised during construction. This allows a certain tolerance of moisture. However, always dry immediately any spillage of water or other liquids inside the kit.

The temperature of the incubator should be checked periodically eg. annually, as indicated in the section on 'Checking and Recalibration of the Incubator' (See Page 52).

Filtration Apparatus

At the end of each day, it is good practice to carefully dry all components of the filtration apparatus including the vacuum and sample cups and to sterilise the apparatus. This practice prevents corrosion of the aluminium components of the filtration apparatus.

Chlorine and pH Comparator and Turbidity Tubes

Avoid scratching the comparator and the turbidity tubes. They rely on an adequate transmission of light for accurate results.

Keep the surfaces clean and dry and free of residues which may prove difficult to remove once dry. After use always wash in clean water. *Never* use detergents, acids or organic solvents.

Kit Case

The outer case is robust and resilient and will withstand a certain amount of harsh treatment. However, try to avoid abrasion and hard impacts.

Maintenance

Weekly:

Apply a smear of silicone grease to the black rubber O-ring.

Wash, rinse and dry the filtration apparatus.

Charge the internal battery fully at the end of each week.

Annually:

Check the incubator temperature and recalibrate if necessary.

Evaluation and Repair of the Kit

Fault Finding in the Incubator, Battery and Charger

1. Connect the battery charger unit to the incubator. Do not switch on the incubator. Connect the charger to the mains electricity supply.

Do the red and green lights on the charger unit light up when the mains supply is switched on?

Yes Go to Step 2.

No Go to Step 6.

2. Charge the internal battery according to the instructions given earlier in the manual.

How many hours does it take from switching on the charger to the point where the green light is permanently extinguished ie. the battery is fully charged?

Note: If after 72 hours the green light is still on, the battery is completely damaged or worn out and will require replacement. Battery replacement should be carried out only by a qualified electronics technician. A battery replacement kit is available from the Robens Institute.

Generally, the total discharge of the battery is a sign of misuse.

Go to Step 3.

3. Prepare the kit for temperature checking and calibration as described in the section "Checking and Recalibration of the Incubator" (See Page 52).

Disconnect the charger from the mains electricity supply.

Disconnect the charger from the incubator unit.

Switch on the incubator unit.

Do the two red lights on the incubator console light up brightly?

Yes Go to Step 4.

No Go to Step 7.

4. Leave the incubator switched on until the temperature reading is stable over a period of at least 30 minutes. The time taken for the incubator to reach this point will depend on the ambient temperature but is usually no more than 3 hours.

Does the incubator hold a temperature of between 43.5 and 44.5 °C?

Yes Go to Step 5.

No Go to Step 10.

5. Does the incubator hold a temperature of between 43.5 and 44.5°C for four incubation cycles of 18 hours each, without needing to recharge the battery?

Note: Between each cycle leave the incubator to cool for at least 8 hours.

Yes Your incubator and battery charger are in good condition.

No Go to Step 9.

6. The charger fuse may have blown.

Ensure that the charger unit is disconnected from the mains electricity supply.

Replace the internal fuse in the charger (see page 51.)

Reconnect the charger to the mains electricity supply.

Do the red and green lights on the charger light up?

Yes Go to Step 2.

No The charger is damaged. Replace with a new unit or organise repair through the Robens Institute or a qualified electronics technician. Then go to Step 1.

Note: The battery charger unit is not a normal battery charger. Always use the correct spare parts, which may be obtained from the Robens Institute. The use of a car battery charger on the equipment will cause permanent damage to the battery.

10. Follow the incubator recalibration procedure given in the section "Checking and Recalibration of the Incubator" (see page 52).

Does the incubator hold a temperature of between 43.5 and 44.5°C after adjustment?

Yes

Go to Step 5.

No

The incubator is damaged. Contact an electronics technician trained by the Robens Institute to carry out repairs or return the kit to the Robens Institute for repair. A Repair kit is available from the Robens Institute.

11. Disconnect the battery charger from the incubator unit. Connect the incubator unit to a well charged 12v battery using the lead with crocodile clips supplied with the kit. Switch on the incubator unit.

Do the two lights on the incubator console light up brightly?

Yes

Go to Step 12

No

The incubator is damaged. Contact an electronics technician trained by the Robens Institute to carry out repairs or return the kit to the Robens Institute for repair. A Repair kit is available from the Robens Institute.

12. Check the reading on the thermometer.

Does the incubator hold a temperature of between 43.5 and 44.5°C?

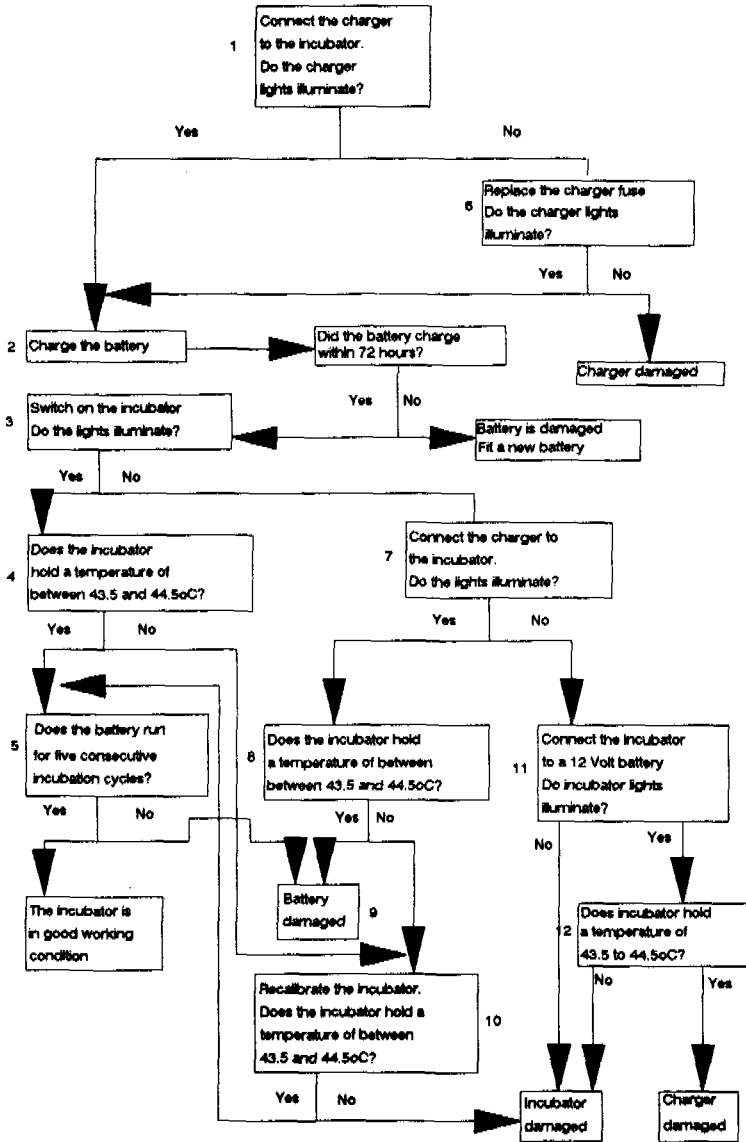
Yes

The charger unit is damaged. Replace with a new unit or repair through the Robens Institute or a qualified electronics technician then go to Step 1.

No

The incubator is damaged. Contact an electronics technician trained by the Robens Institute to carry out repairs or return the kit to the Robens Institute for repair. A Repair kit is available from the Robens Institute.

Fault Finding Chart



A damaged or worn out kit can only be repaired by the Robens Institute if a funding agency will accept the costs involved ie. repair, freight and insurance.

When you return your kit please send a letter guaranteeing payment.

Send kits for repair to the address at the front of this manual.

Before sending your kit please remove all loose items from the box such as the turbidity tubes, chlorine tester, filtration apparatus, etc. These items may be lost or damaged in transit and will increase freight costs.

Replacing the Charger Fuse

To replace the charger fuse you will need the following items:

Spare fuse. Spare fuses are supplied with new kits in the spares box.

Flat blade screwdriver (the tweezers supplied with the kit can be used if a screwdriver is not available).

Procedure for Replacing Charger Fuse

- a) Ensure that the charger unit is not connected to the mains supply or the kit.
- b) Place the charger on a firm surface with the indicator lights facing you.
- c) Unscrew the four recessed plastic bolts at the corners of the charger using a screwdriver or the points of the tweezers.
- d) Carefully pull off the lid of the charger. As the charger unit is designed to be showerproof, the lid may be quite tight and it may help to break the seal by carefully inserting one point of the tweezers between the lid and the charger box.

Take care not to lose the four plastic bolts which may fall out when the lid is removed.

- e) The fuse is situated on the rear left hand side of the charger. Prise up one end of the fuse with a screwdriver or tweezers and lift the fuse out of its spring clips.
- f) Place the new fuse onto the two spring clips and press into place with a finger.
- g) Replace the charger unit lid and tighten the four plastic bolts with a screwdriver or tweezers. Return to step 6. of the fault finding guide.

Checking and Recalibration of the Incubator

The equipment supplied for checking and recalibration of the incubator includes the following items. Please check them before use.

Testing lid with centre hole

Thermometer

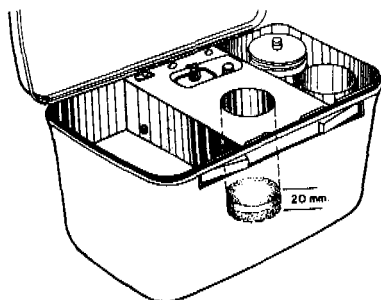
Trimmer tool (similar to a small screwdriver)

Note: It is recommended that the temperature of the incubator is checked once every 12 months.

Procedure for Checking the Incubator Temperature

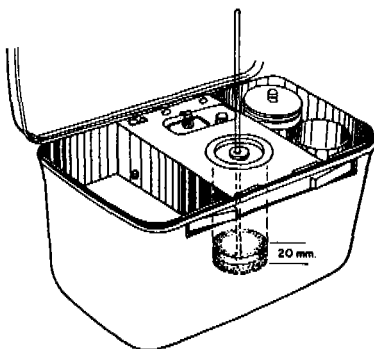
a) Remove all contents from the kit and wipe clean the internal surfaces with a clean, damp cloth or paper towel.

Pour approximately 20ml of clean water into the incubator pot.



b) Push the thermometer through the hole in the testing lid.

c) Replace the incubator lid with the testing lid and thermometer assembly, allowing the bulb of the thermometer to be completely immersed in the water.



Carry out the following procedure at an ambient temperature of between 15°C and 25°C.

d) After ensuring that the internal battery is completely charged or that the kit is operating from a mains electricity supply or well-charged external 12V battery, switch on the incubator.

e) Check the temperature of the incubator and observe over a period of 30 minutes to make sure that it has stabilised. The incubator normally takes no more than 3 hours to reach a stable temperature, depending on the ambient temperature.

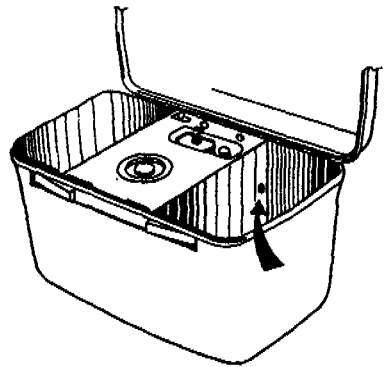
f) If, once the incubator has stabilised, the temperature is between 43.5 and 44.5°C then recalibration is not necessary. If however, the temperature is not within these limits, follow the recalibration procedure below.

Procedure for Recalibration of the Incubator

g) Leave the testing lid and thermometer assembly in place and keep the incubator switched on.

h) Peel off the small black triangle situated on the right hand side of the incubator console and keep it in a safe place.

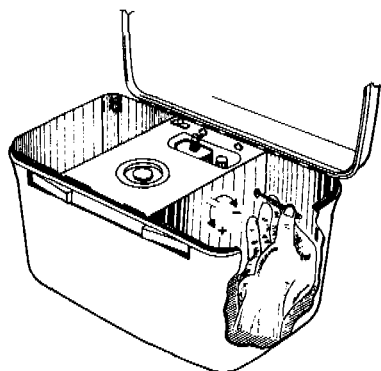
Insert the trimmer tool into the hole beneath the black triangle and locate the tool in the calibration screw.



Note: Small adjustments of the screw make large adjustments to the temperature. A quarter turn (90°) results in a temperature change of approximately 1°C.

i) To increase the temperature, turn the adjustment screw anti-clockwise (+). To decrease the temperature, turn the adjustment screw clockwise (-). Make any adjustments in stages, a little at a time. After each adjustment, leave the incubator to stabilise for at least 30 minutes.

The complete recalibration procedure may take several hours. Be patient.



j) Once the incubator has been recalibrated to read between 43.5 and 44.5°C, leave switched on for at least 3 hours. Take note of the temperature at 30 minute intervals to ensure that the temperature is stable.

k) Switch off the incubator and leave to cool. Do not disconnect the incubator from the mains electricity supply.

l) The following day, switch on the incubator and allow to reach a stable temperature. If the temperature is not within the correct limit repeat the checking process detailed in steps (g) to (j).

m) Dismantle the temperature checking equipment and store in a safe place.

n) Replace the black triangle over the calibration screw hole.

Note: The procedure above guarantees an average incubator temperature +/- 0.5°C. The temperature in the incubator may vary within +/- 0.5°C during incubation after reaching the set temperature.

Appendices

Daily Report Sheet

Health Region _____ Authority _____

Date ____/____/____ Code _____

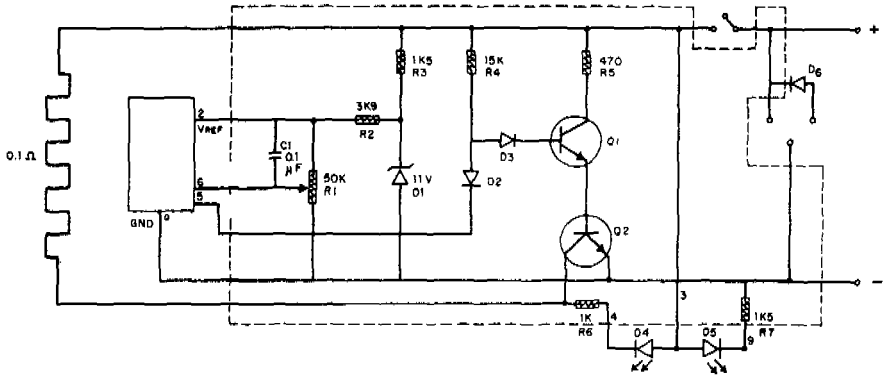
Province _____ District _____

Community _____ Analyst _____

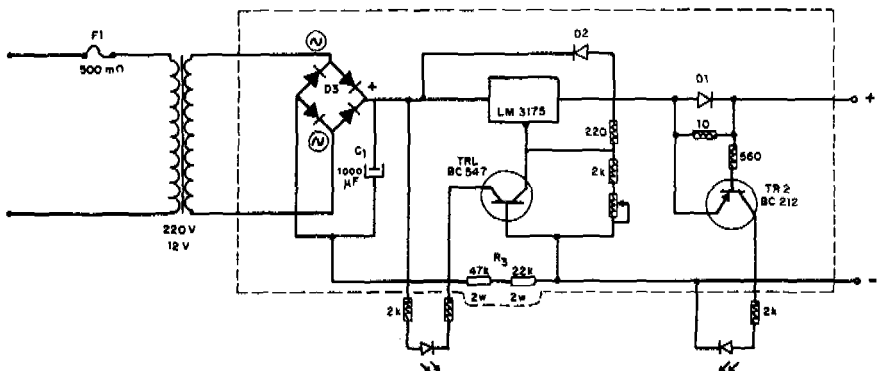
| Source of Sample | Time | Colour | Odour | Turbidity NTU | Chlorine (Mg/L) | | | pH | Faecal Coli (F.C) | | |
|------------------|------|--------|-------|---------------|-----------------|-----------|------------|----|-------------------|------------|-------------|
| | | | | | Free Mg/L | Comb Mg/L | Total Mg/L | | Vol filt ml | No. of col | F.C/ 100 ml |
| | | | | | | | | | | | |
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Electronic Circuit Diagrams

Incubator



Charger



Fieldwork Checklist

Before leaving for the field, check that you have the following items:

Kit

Filtration apparatus
Sample cup
Vacuum cup
Sample cable
Vacuum pump
Turbidity tubes (pair)
Chlorine/pH comparator
Tweezers
Petri dishes
Lighter
Spares box

Consumables

Culture medium
Membrane filters
Absorbent pads and dispenser
DPD No. 1 tablets
DPD No. 3 tablets
Phenol Red tablets
Methanol
Report Sheets
Paper towels or clean cloth

Spares List

The following spares and consumables are available from the Robens Institute. Please telephone, fax or write for a current price list.

Component

Battery replacement kit containing:

- Battery 12v 9.5Ah
- Silicone sealant

Temperature check kit containing:

- Testing lid
- Thermometer
- Adjuster tool
- Charger fuses

Electrical repair kit containing:

- Electronic circuit
- Temperature chip
- Sealant
- Foam compound
- Adhesives

Filtration apparatus (complete)

Filter funnel with plastic collar

Vacuum cup

Sample cup

Filtration base

Vacuum pump

Sample cable

Bronze disc with silicone rings

Black rubber O-ring

Turbidity tubes (pair)

Spares box (empty)

Forceps

Component

Lighter
Chlorine/pH comparator
External battery cable
Silicone grease (2g)
Charger fuses (2)
Polypropylene bottles 60ml (10)
Methanol dispenser

Consumables

Membrane filters with pads (200)
Membrane filters with pads (1000)
Culture medium 38.1g tub for 500ml of growth medium
(sufficient for 200 tests)
Culture medium (500g tub for 6.5 litres of growth
medium (sufficient for 2,600 tests)
DPD No.1 tablets (250)
DPD No.1 tablets (1000)
DPD No.3 tablets (250)
DPD No.3 tablets (1000)
Phenol red tablets (250)
Phenol Red tablets (1000)

Overseas Development
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Notes

Notes



World Health Organization Collaborating Centre
for the promotion of Drinking Water
Quality and Human Health

Overseas Development

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