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MAINTAINING CONCENTRATION: A NEW PRACTICAL METHOD FOR PROFILING AND TOPPING UP ALCOHOL-PRESERVED COLLECTIONS

David G. Notton

Department of Entomology, the Natural History Museum, London, SW7 5BD, United Kingdom, d.notton@nhm.ac.uk

Abstract.—A new method of profiling alcohol-preserved collections is presented and its use as a diagnostic tool is discussed. Some previous methods for topping up are reviewed and a new method is proposed. A novel tool is presented—a reference table for calculating the concentration of topping-up alcohol—which allows the regulation of preservative alcohol concentration within close limits. The method can be used for remedial and routine topping up and can be adapted to the needs of different collections.

Introduction

The first line of defence against evaporation of fluid preservatives always must be the best storage jar affordable; however, all storage jars allow at least some evaporation and need a regular schedule of inspection, maintenance and, topping up. How to top up effectively is the subject of this paper.

A new method of profiling alcohol-preserved collections is presented which takes into account the volume of preservative present, and the use of profiling as a diagnostic tool is discussed. Some previous methods for topping up and their useful features and limitations are considered. A new approach is taken to topping up, which allows the desired concentration to be achieved while taking into account any variation in starting concentrations and volumes. The new topping-up method has a number of key features. By analogy, with the control of temperature and humidity for purposes of conservation, potentially damaging fluctuations are managed much more closely, both in alcohol concentration and in volume. Greater weight is given to the volume of preservative present than previous methods for a number of reasons, not only to prevent the specimen becoming exposed, but also because it provides an indicator of low alcohol concentration, because low volumes can be an indication of a faulty seal, and because it is important in calculating the correct concentration for topping up. The novel tool presented is a reference table for calculating the concentration of topping-up alcohol to add, which makes the method applicable to large collections. The table gives speed and convenience priority over accuracy; however, because of the use of precise monitoring, the results are still accurate within close limits.

A general approach is developed which can be applied to many alcohol-preserved collections. The methods proposed are designed for use with collections in modern storage jars, or where replacement and standardisation of jars is possible. Unless specified, the term alcohol is used here to include ethanol and also the mixed alcohols (ethanol and a small percentage of methanol used as a denaturant) found in industrial methylated spirits (IMS), although the properties of IMS differ slightly from pure ethanol solutions. Alcohol concentration was measured using the Anton Paar DMA 35N digital alcohol meter, www.anton-paar.com. This meter was used on the "%ALC/V" setting, which gives a measurement of the equivalent concentration of an ethanol/water mixture in percent by volume at 20°C derived from density at the measuring temperature (Anton Paar 2000). The methods should work equally well where propylene glycol (1, 2

propandiol) is used as a humectant because this has a negligible effect on density measurements (Boase and Waller 1994). However, the methods should be applied with caution where it is suspected that other humectants, fixatives, and buffers are present that could affect density measurements. The application of the method in cases of acidification, leaching, and large concentration changes, is considered. For best results, the environmental conditions within a storage area should be managed closely, although the method can be adapted to some extent for the different conditions in stores. Methods appropriate to collections with historic jars, which might need to be conserved in their own right, are not considered in detail. The methods described are not intended for use where the preservation of DNA is of primary importance. It is not the purpose of this paper to consider the initial preservation of specimens, and it is assumed that specimens are preserved and equilibrated with their preserving fluid. The mention of any brand names in this paper does not constitute an endorsement by the Natural History Museum.

This paper is written from experience of working with the large and varied collection of entomological and other terrestrial arthropods preserved in IMS at the Natural History Museum, London. It includes a wide range of taxa and sizes of specimens from large scorpions and myriapods (c. 20 cm) to tiny insects (c. 1 mm) (but not including Crustacea) in a range of jar types, including historic ground glass in a variety of sizes, Bakelite- and polyethylene-topped jars, metal-topped jam/honey jars, and the newest standard ''Le Parfait'' bail-top jars. Smaller specimens often are contained in glass vials, with old cotton wool or newer polyethylene stoppers, which allow many vials to be kept together in one jar. This collection only recently has been brought together from a variety of locations, and has been kept together since 2005 in the purpose-built Darwin Centre spirit store.

What Happens When Alcohol Evaporates and the Consequences for Topping Up

Evaporation from alcohol-based preservatives is affected by: the temperature of the air–liquid interface, the concentration of vapours above the surface, the area of the vapour–liquid interface, and the partial vapour pressures of the different components (Simmons 1991, 1995a). In the case of predominantly ethanol/water mixtures normally used as preservatives, the ethanol generally evaporates faster than the water because the vapour pressure is higher than that of water (Simmons 1995a; Waller and Strang 1996), and evaporation from ethanol/water mixtures is generally faster than might be expected from the evaporation rates of water and ethanol on their own (positive deviation from Raoult's law). In alcohol-preserved collections, these effects are countered by maintaining low temperatures and enclosing specimens in sealed jars, which maintain high vapour concentrations above the liquid, preventing more evaporation. Maintaining high vapour concentrations inside the jar depend on providing a good seal which minimises the vapour escaping, and having a relatively impermeable jar. The conditions in the store also are important (Simmons 1991; Horie 1994); for example, if the relative humidity is high but the alcohol vapour pressure is low, proportionally more alcohol will evaporate than water, further reducing the concentration of alcohol in the jar over time. If the relative humidity is particularly high, ethanol/water mixtures are sufficiently hygroscopic to absorb water from the atmosphere. Eventually, enough ethanol will escape, the concentration and volume of the preservative will drop, and the container will need to be topped up. This presents a problem for topping up, in trying to return the jars to the correct concentration, because a jar containing a lot of weak alcohol will need stronger alcohol to be added than a jar containing the same amount of strong alcohol. Waller and

Strang (1996) explained much of the underlying physical chemistry and provided some useful examples of how alcohol is lost; e.g., a 70% ethanol/water solution evaporating into air of 0% relative humidity would lose vapour equivalent to 87% ethanol if condensed. Similarly, a starting solution of 60% would lose vapour equivalent to 86% ethanol, and a starting solution of 80% would lose vapour equivalent to 89% ethanol. Because most stores have more than 0% relative humidity, 87% alcohol would be about the minimum concentration that could be used to top up if any attempt is made to maintain concentration. In a real store at a not-untypical 50% relative humidity, a 70% ethanol/water solution would lose vapour equivalent to 95% ethanol if condensed, and would need to be topped up with correspondingly strong alcohol (van Dyke 1980; Waller and Strang 1996), much stronger than often is recommended.

The Aims of Topping Up

The aims of topping up are:

- To keep the specimen covered with preservative, so: a) it does not dry out, b) it is physically supported, and c) evaporation does not lead to the deposition of salts and other solutes on the exposed portion of the specimen.
- To maintain the correct concentration of alcohol for preservation. For insects, this generally is considered to be 70–80% for general preservation (not for DNA, however, which needs higher concentrations). Lower concentrations can cause distortion of the specimen by absorption of water and autolysis. Concentrations of 50–80% are recommended by Waller and Strang (1996) as the range with the best antiseptic properties; below 50%, growth of bacteria and mould become increasingly likely. Higher concentrations can cause tissue distortion and embrittlement.
- To keep fluctuations in concentration limited within acceptable boundaries, by analogy with other methods of environmental control. Osmotic pressure increases particularly rapidly for concentrations above 80%, but also rises steadily between concentrations of 0–75%, suggesting that large changes in concentration during topping up should be avoided as a precaution against osmotic stress, which can distort the specimen (Waller and Strang 1996).

There still is debate about the best alcohol concentrations for preservation and the acceptable limits of fluctuation in concentration for insects and other natural history specimens (Table 1; Levi 1966). It is not the purpose of this paper to decide what these precise values are, but to address the more general question of how to maintain a given concentration within given limits of fluctuation. For the purpose of this paper, the desirable concentration is arbitrarily considered to be 75% and the acceptable limits of fluctuation in concentration are set at ±5%. The acceptable limits to fluctuation are set narrowly, on the precautionary principle that there is very little information on acceptable risk levels for alcohol concentration and the effects of changes in concentration are not well understood (Cato 1990; Pickering 1997). Careful comparisons of different techniques are needed; e.g., a comparison of topping-up method for spiders (Cushing and Slowik 2007) is of interest. However, it is difficult to make generalisations from this paper because it was based on specimens whose history suggested they were already degraded, and no microscopic assessment of tissue quality was made. Much remains to be understood about the causes of deterioration of alcohol-preserved specimens (Hancock 1985; Wallace 1985; Noyes 1990; Simmons 1991; Morse 1992; Masner 1994) and how this relates to the concentration and fluctuations in concentration

Table 1. Some published methods for topping up alcohol-preserved collections. IMS = industrial methylated spirits.

Author	Scope of collection	Starting concentration	Inspection	Concentration monitoring	Addition/discarding of preservative	Topping-up records
Van Dam 2002, 2008	Human anatomy	70–80% ethanol	Yes	Coloured plastic indicator beads permanently kept in container. Beads float when fluid concentration is below 60% ±3% (orange disc) and below 50% ±3% (red disc)	If concentration 50–60%: if fluid volume ≥ 50%, discard fluid until volume is 50%, then top up with 96% ethanol; if volume is 33–50%, top up with 90% ethanol; if volume is 10–33%, top up with 80% ethanol.	?
Moore 1999	Natural history	70–90% IMS	Monitor periodically	DMA 35 digital density meter or hydrometer	Check strength of preservative; top up with "correct" fluid; replace preservative contaminated by lipid leaching; replace gasket or lid if needed; grease around ground-glass lids.	Label on back of jar, ledger and photos
Waller and Strang 1996	Biological	70–75% ethanol	Yes	Yes	Top up with 87–95% ethanol, depending on relative humidity in store.	?
Sendall and Hughes 1996	Fishes in vats	45 or 60% (?isopropanol)	Yes	Yes	Formula based on desired end concentration, height of fluid in vat, concentration of fluid in vat, and concentration of fluid being added, to give the height of fluid to add.	?
Simmons 1995a, 1995b	Natural history	70% ethanol or denatured ethanol	Yes	Digital density meter or hydrometer	Concentration of fluids in container should be measured to determine what should be added to achieve the desired solution.	?

Table 1. Continued.

Author	Scope of collection	Starting concentration	Inspection	Concentration monitoring	Addition/discarding of preservative	Topping-up records
Carter, 1994, 1995	Natural history	80% IMS	Yes	DMA 35 digital density meter, specific gravity readings converted into alcohol concentration using programme devised by T. Strang	Top up with 80%, and after two topping-up operations where > 40% of preservative volume is lost each time, replace all fluid.	Yes
Reilly 1989	Animals, including invertebrates	70% ethanol or 70% + 5% glycerine	Periodically, 1–2 years	Hydrometer	Measure strength of remaining fluid and "top up accordingly."	?
Walker and Crosby 1988	Insects	75% ethanol	Every 2 years	?	Add 95% Ethanol.	Last topping up date displayed in store
Van Dyke 1980	Fish	75% ethanol	Yes	Hydrometer	Add 87–88% minimum.	?

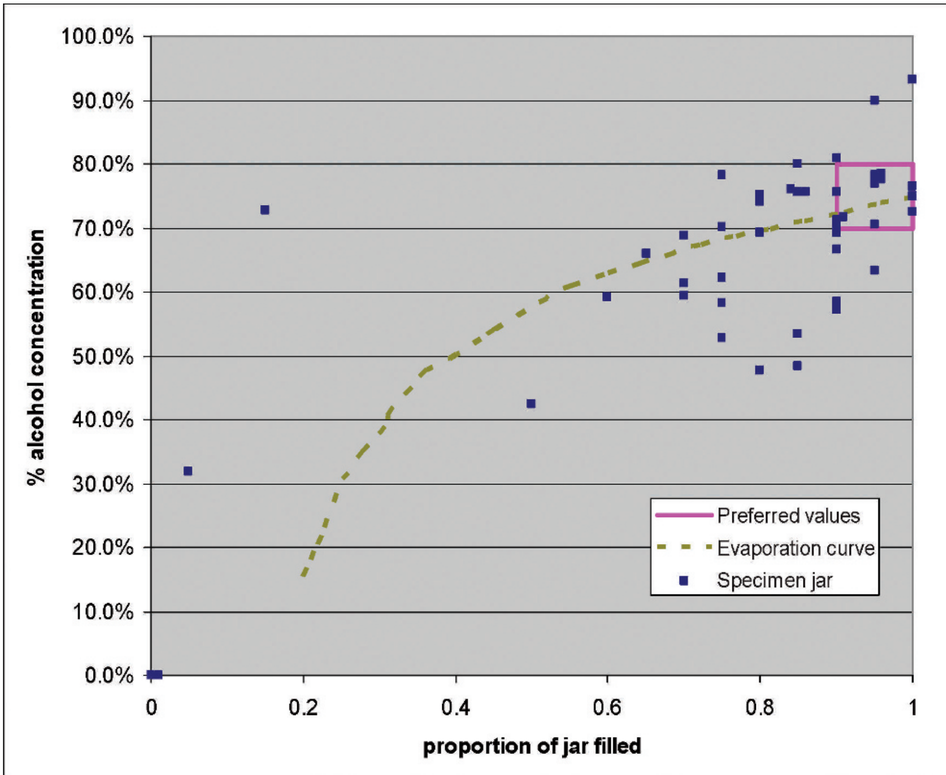

Figure 1. Profile of Entomology Department alcohol-preserved collection, August 2007. Each square represents data for a jar in the collection. The evaporation curve was obtained from a beaker of 75% IMS in the Darwin Centre store, which had been allowed to dry out without topping up (data from Protocol 1, page 17).

of alcohol. Although the method below is based on these values, it is easily adaptable to others.

Collection Profiles—Taking Account of Volume As Well As Concentration Helps to Diagnose Problems with Topping-up Procedures

Before starting to top up a large collection, it is advisable to make a profile of the collection (a snapshot of its condition), first to find if the aims of topping up are being met, so treatment can address any problems, and secondly to estimate the materials needed. Previous collection profiles have presented concentration as a frequency histogram (Cato 1990; Pickering 1997); although this is a very useful and straightforward approach, it records no information about the volume of preservative present. Instead, a new method was used instead, which plotted concentration of alcohol against the volume of preservative (as a proportion of the jar filled). This allowed assessment of the profile of concentrations as was done by Cato and Pickering, but also assessment of the profile of volumes and any interaction between concentration and volume; e.g., if the preservative is more dilute than expected from its volume. An advantage of this approach is that a "target area" of acceptable concentration and volume can be superimposed on the graph and the proportion of the collection in the target area counted and used as an indicator of "collection health."

Before developing new topping-up methods for the Natural History Museum entomology spirit collection, a profile was made (Fig. 1). Two jars were selected from

each row of cabinets in the store, including samples of all the common jar types present. For each jar, the proportion filled with preservative was estimated and the alcohol concentration measured. A designation of "100% full" is defined as filled up to the neck, leaving a small head space of c. 10 mm. A simple evaporation line was also added, from a beaker of 75% IMS in the Darwin Centre store, which had been allowed to dry out without topping up (data from Protocol 1, page 17).

The graph showed that:

- Topping up had not been applied consistently, as shown by the jars not filled or not having the right concentration.
- Many jars were not filled adequately; probably they were not been topped up fully or there was a previously unrecognised problem with the seal. Some specimens that were not double-housed were at risk of drying and suffering damage where they were exposed above the preservative.
- Many jars were at the wrong concentration: about a third were too low, and surprisingly about a fifth were more concentrated than expected, some considerably more concentrated. Clearly there was a problem with topping up, because an alcohol meter has been available to all staff since 2002. For jars at low concentration, especially those <60%, action needs to be taken soon as there is a serious risk of autolysis, microbiological decomposition, and distortion. Enquiries regarding those jars at high concentration (>80%) found in one part of the collection, showed that these were part of a group of vials bottled up recently, and some strong alcohol had been used in error.
- Many jars in the area below the sample evaporation curve were less concentrated than might be expected for their volume. In all probability they were topped up with under-strength alcohol (probably 80%) which is known to reduce concentration over time.

Although there is apparently widespread awareness of the importance of topping up and of monitoring concentration, there is some way to go to improve both volume and concentration of alcohol. Problem areas of the collection need to be identified in more detail and prioritised for remedial topping up. Better routine concentration monitoring and topping up is needed generally.

The 80% Fallacy—Some Previous Topping-Up Methods and Their Limitations

It has been recognised amongst biology curators (e.g., Harris 1977; Carter 1995; van Dam 2002) that the concentration of alcohol solutions changes on evaporation, and that testing might be needed before topping up, so that topping up returns the alcohol concentration to a sufficient strength. Some rules of thumb, e.g., repeated topping up with 75–80% alcohol can, over time, cause a steady decrease in concentration (Cato 1990; Carter 1995), and as mentioned above, 87–96% are more realistic concentrations for topping up alcohol if any attempt is made to maintain concentration (van Dyke 1980; Waller and Strang 1996). Even so, there is little agreement over the best method for topping up alcohol-preserved collections, and specifically those for terrestrial arthropods. In many cases, the methods presented were sketchy on details of, and reasons for, the alcohol concentration used for topping up. A selection of recently published methods for terrestrial arthropod and other collections is presented in Table 1.

Inspection

Inspection is a vital part of the process of maintaining alcohol-preserved collections, yet no methods gave reasons for the timing of inspections, apart from constraints due to

limited resources (Reilly 1989). The ideal timing for inspections should relate to some measure of the increasing risk of damage to the specimens over time. Van Dam's method which involves inspection as an integral part of concentration monitoring is discussed below.

Measuring Alcohol Concentrations

Three main methods of monitoring alcohol concentrations have been used in recent methods, hydrometers, plastic beads, and digital density meters. Because all methods rely on density determination, they will not work accurately in solutions containing substances with an effect on the solution density, e.g., salts or glycerol.

Float hydrometers have traditionally been used to measure the density of alcohol solutions; they work on the principle of displacement, so the weight of the free-floating hydrometer equals the weight of preservative displaced by it. Unfortunately there is often insufficient preservative in smaller jars to float some hydrometers (Cato 1990). For the adept, small, simple densitometers can be made based on the Galileo thermometer principle; e.g., Moore (1999) used a pipette containing weighted plastic beads to distinguish solutions of different density. A problem with float hydrometers is that they measure density, not concentration, and because of the expansion of alcohol solutions with temperature, density readings need to be corrected to volume % or weight % at 20°C using a conversion table, so that the results are comparable. For this reason float hydrometers calibrated with scales showing concentration, often referred to as "alcoholometers," only give accurate concentration readings at the given reference temperature (usually 20°C). A free computer program, for calculating volume percentages of ethanol (and isopropanol) from densities at given temperatures no longer is available (Canadian Conservation Institute 1994), although a more advanced programme for blending and density concentration conversions for ethanol/water can be purchased (Katmar Software 2009), and standard chemistry texts can be consulted. Individual calculations can be impractical for the large numbers of jars often found in invertebrate collections.

Plastic beads have been used by van Dam (2002) for a historic anatomy collection, to regulate preservative concentrations within the aseptic range 50–80%. Beads of different densities were placed in specimen jars; for each kind of bead the density is such that when the preservative density drops to a certain value, the bead floats, showing that attention is needed. They have the advantage over other measuring systems that the jar need not be opened. Two kinds of beads are used, which float at densities approximating to concentrations of <60% and <50%. The beads can become trapped under specimens, particularly a problem for invertebrate collections where there are many small specimens; to overcome this, the beads must be put in a glass vial closed with a perforated cap and placed inside the specimen jar, increasing the installation time and cost. Although the beads are said to measure concentration, they measure density ranges. The conversion from density ranges to concentration is approximate because there is no temperature correction for temperature-related density changes in the alcohol. Hence, the three possible concentration readings are a) more than 60% ± 3%, b) between 60% ± 3% and 50% ± 3%, and c) less than 50% ± 3% (van Dam undated). Readings taken at different temperatures are only approximately comparable. There remains a small concern over the stability of the beads, and although they are very stable in ethanol/water solutions, van Dam recommends checking a random sample every 10 years. Costs of beads vary with the number purchased; e.g., for a collection of 1,000 jars with 2 beads per jar, beads

€0.50 each would be €1,000 + tax, whereas a collection of 10,000 jars, with 2 beads/jar, beads €0.35 each would be €7,000 plus tax (Alcomon price list for first quarter 2008, www.alcomon.com).

Modern portable digital densitometers are easy to use and can measure small quantities (a few millilitres) accurately and quickly. The hand-held DMA 35N meter used for this project contains a sensor in the measuring cell that measures the sample temperature; this can be used for automatic temperature compensation of the density reading. Therefore, unlike all the other methods above, the display options %ALC/W or %ALC/V units give the alcohol concentration by weight or by volume, automatically corrected to 20°C. All readings are accurate and directly comparable, irrespective of the temperature at which they were taken, without the need for time-consuming temperature conversion tables (Anton Paar 2000). Such meters effectively overcome all the problems of float hydrometers. Compared to the plastic bead system they 1) are much more accurate, 2) provide temperature-corrected concentration readings, 3) allow much finer monitoring and control, 4) are not tied to measuring predetermined density ranges, and 5) do not require the time to install the beads. However, they are slightly more time-consuming per measurement, and the jar needs to be opened. The cost of a digital density meter is significant but is cost effective for large collections (DMA 35N digital density meter €1660, plus tax, June 2008).

Adding and Discarding Preservative

For those authors giving explicit methods, two main strategies can be identified: 1) topping up with sufficiently strong alcohol to maintain the desired concentration; and 2) topping up with under-strength alcohol (80%), accepting that the concentration in the jar will decrease over time, and when it decreases to a certain level, all the preservative will need to be replaced with fresh alcohol of the correct concentration. These and the other methods differ also in the levels of fluctuation in alcohol concentration and volume.

A major problem with previous methods belonging to the first strategy is that most ignore the problem of how to choose the correct concentration of alcohol to add to return a dilute solution to the desired concentration. A useful approach to this problem was made by Sendall and Hughes (1996), who gave an equation based on the desired end concentration, height of preservative in the container, concentration of alcohol in the container, and concentration of fluid being added, to give the height of preservative to add. This was intended for topping up vats of fish, and although effective, is time consuming to work out for more than a few containers, and the calculation does not assume the vat is to be filled. Van Dam (2002) also addressed the problem; he noted three different concentrations that should be used when topping up when starting from different volumes and under some common scenarios found when using his method. Unfortunately these are not subtle enough for close regulation of concentration and volumes because the method of measuring the initial volume and concentration of alcohol present is imprecise. Hence a general-purpose algorithm is needed, which can make these calculations accurately and quickly and is suitable for use with large collections.

For the second strategy, topping up is done with under-strength alcohol, usually 80%, accepting that the concentration in the jar will decrease over time, and when the volume decreases significantly, all the preservative will need to be replaced with fresh alcohol of the correct concentration (e.g., Leis and McGrouther 1994; Carter 1995). This sometimes is used for vertebrate collections and has the advantage that it reduces the risk of damage

to the specimen through acidification of the preservative from the breakdown of lipids into fatty acids. It also is simple because it uses one strength of alcohol. For terrestrial invertebrate collections, however, the specimens generally are smaller in relation to the volume of preservative, the amounts of fat are relatively smaller, and the observed frequency of lipid leakage is much lower (as judged by discoloration, cloudiness, and obvious oil globules, although the quantification of this problem by pH monitoring has yet to be undertaken). Acidification is less likely to be a problem for invertebrates, and the benefits of replacing all the preservative are less clear and need to be balanced against a number of other factors: 1) increased leaching of components of the specimen, 2) the cost of the replacement preservative, 3) the cost and time taken for safe disposal of the old preservative as chemical waste, and 4) environmental pollution caused by disposal of old preservative.

The last two points have become more important in recent years; e.g., in support of its environmental accreditation the Natural History Museum now aims to collect and dispose of waste preservative as chemical waste because this is considered less environmentally damaging than discharging it into the public sewerage system. However, disposal of chemical waste is costly, so the minimisation of this kind of waste is desirable.

Compared to replacing all the alcohol with 80%, retaining the original alcohol and topping up with a smaller amount of strong alcohol is comparable in price. This is because although 96% IMS is about twice the price of 80%, only about half the volume of 96% is needed, because the original alcohol is not discarded. For example, 1 L of 80% IMS costs £0.54–1.19 + VAT, whereas 500 ml of IMS 96 costs £0.37–1.41 + VAT (Hayman, Ltd. prices quoted 20 May 2008; exact prices depend on quantity). Thus, topping up with a smaller amount of stronger alcohol is better where acidification is not a problem because it reduces leaching, is usually about the same cost or cheaper for the price of the topping-up alcohol, and saves the cost of disposal and pollution of the environment with contaminated waste preservative. It only is recommended that all preservative is replaced where there is evidence of acidification (pH monitoring of alcohol solutions is not straightforward, but cannot be dealt with here). Discoloration, however, is not a certain indicator of acidification. Taxa known to be at risk should be checked more regularly.

There are differences in the levels of fluctuation of alcohol concentration and preservative level allowed by the different methods. For example, van Dam (2002) allowed fluctuation from 75% to somewhere in the range 60–50% ± 3%, arguing that this is acceptable because it maintains an antiseptic range of 80–50%, based on Waller and Strang (1996). It is possible, however, that such a large change in concentration and osmotic pressure likely can lead to soft tissue damage. Reid (1994) advocated the use of extended-step sequences to avoid osmotic damage, and Waller and Strang (1996) recommended that solutions with approximately equal concentration increments for stepping specimens up to higher ethanol concentrations are used up to about 80%. They did not establish what the desirable step size is, but the single steps of c. 12–28% (from 60–50% ± 3% back to c. 75%) during topping up implied by van Dam's (c. 2008) method seem large, and should be qualified to say that the topping up should be done in stages. Very small osmotic pressure changes can lead to shrinkage of some organisms, so small steps are advocated on a precautionary principle that little is known about the effects of osmotic shock on long-term invertebrate tissue preservation. The reduction of preservative volumes is an associated problem; it appears that van Dam (2002) was prepared to tolerate exposure of the specimen above the alcohol for a limited period, at least "as long as the fluid and its saturated vapor provide in sufficient antiseptic protection, fluid loss does not have to result in immediate damage to the [s]pecimens [sic]." It is reasonable to suppose, however, that when a

specimen is exposed, van Dam would top up the jar, even when neither of the beads are floating (although no mention is made of how to determine the concentration of alcohol to add in this situation). Generally, it is best to avoid such low levels altogether because low levels could indicate a faulty seal and possible imminent failure and irreparable damage to the specimen—allowing levels to go low would provide less time for remedial action. Specimens should not be allowed to become exposed because the preservative does not provide mechanical support for fragile specimens; and evaporation from the exposed part of the specimen can cause encrustation of solutes. Hence, it is recommended that fluctuations in volume and concentration are minimised as far as possible; a parallel is made with other methods of environmental control (temperature and relative humidity) where it is generally accepted that minimising fluctuations is desirable.

Records

Little has been published about keeping records of topping up, but it should be useful in 1) evaluating the effectiveness of topping-up methods, 2) determining performance of jars, 3) recording conservation history of specimens as a management tool, and 4) providing evidence for collections development proposals. Moore (1999), a professional conservator, is noteworthy as one of the few workers recommending record keeping, using a label on the specimen jar, a ledger, and photographs if required. Thought needs to be given to how records are kept and how they are linked. Linking records to specimen records can provide a history of the treatment of specimens, but might not be practical where 1,000s of small specimens are kept. Alternatively linking records to jar numbers could provide a record of the effectiveness of jars. Both ways of linking conservation treatments are possible in modern relational databases. The recording of environmental variables for a store (especially relative humidity and temperature, both of which have direct effects on evaporation rates), is also desirable. This aspect of topping up will not be explored further here except to highlight its importance.

Conclusions

For the reasons above, although many of the previously published methods have useful features, none is entirely adequate for terrestrial invertebrate collections. Aspects of good practice can be combined together as follows:

- Checking of alcohol concentration should be done systematically (not randomly), especially where remedial topping up is concerned (Cato 1990).
- Concentration should be measured using a rapid digital density meter with automatic correction to standardised volume % at 20°C, because this is much more accurate than any other method.
- To avoid leaching, complete replacement of preservative should be used only for specimens likely to be at risk of acidification, e.g., fatty larvae, large beetles, and galls, or where there are chemical contaminants such as buffers, humectants, and fixatives, which can be hazardous for staff or damaging to specimens.
- A range of alcohol concentrations (often much more concentrated than past conventions would suggest) should be used for topping up so as to return concentration to the desired value.
- As a precaution against osmotic stress, interventions should be made sooner, before a large proportion of alcohol has evaporated, and step changes in alcohol concentration during topping up should be minimised.

The following aspects demand further study and development:

- A quick and practical algorithm for calculating the concentration of topping-up alcohol to add to each jar.
- A better assessment should be made of the timing of inspection needed, based on an empirical assessment of the risk of damage to specimens from being exposed above the preservative surface and due to fluctuation in alcohol concentration.
- Further investigation of the risks of acidification, bleaching, and the performance of vials and closures.

The following section further develops the first of these three ideas.

Developing Practical Methods for Topping Up: A New Algorithm Presented as a Handy Topping-Up Table

It has been explained above that, because of the different treatments and conditions that specimens jars might have undergone, different concentrations of topping-up alcohol might need to be added to restore them to the desired concentration and volume. There is, however, no practical algorithm published, suitable for use with large collections and adaptable to the needs of different users, that would allow calculation of the concentration of alcohol to be added in a reasonable time. The closest approach to date to this problem was by Sendall and Hughes (1996) who gave a formula based on desired end concentration (z), height of fluid in vat (x), concentration of fluid in vat (a), and concentration of fluid being added (b), to give the height of fluid to add (y):

$$z(x+y)=ax+by$$

This method was intended for topping up large vats of fish. Because calculations must be worked individually, its application to large numbers of jars would be troublesome. However, it should be seen as an important advance and provides the basis for the present method. Sendall and Hughes' equation can be modified as follows: for adding two volumes of alcohol of given concentrations, the following approximation can be made (approximation due to slight volume changes on mixing; initial concentration of alcohol = c_i; final concentration of alcohol = c_f; concentration of alcohol added = c_a; initial volume of preservative = v_i; and volume of preservative added = v_a):

$$c_f(v_i+v_a)\approx c_i v_i+c_a v_a$$

The equation can be simplified by dividing by $(v_i + v_a)$, converting volumes to proportions, and substituting V_a to reduce the number of variables to four (initial volume of preservative as proportion of final volume of preservative $v_i/(v_i + v_a) = V_i$; the volume of preservative added as proportion of final volume of preservative $v_a/(v_i + v_a) = V_a$; $1 = (V_i + V_a)$:

$$c_f(v_i+v_a)/(v_i+v_a)\approx c_i v_i/(v_i+v_a)+c_a v_a/(v_i+v_a)$$

$$c_f\approx c_i V_i+c_a V_a$$

$$c_f\approx c_i V_i+c_a(1-V_i)$$

Rearranging gives the concentration of alcohol to be added, expressed in terms of the initial concentration, the final (desired) concentration, and the proportion of the jar

initially filled with preservative:

$$c_a(1-V_i) \approx c_f - c_i V_i$$

$$c_a \approx (c_f - c_i V_i)/(1-V_i)$$

The benefits of this approach are: firstly, proportions can be used rather than absolute measurements of the height of fluid in the jar; hence, the method can be used with any size of container. Secondly, the equation is simplified by assuming the container will be topped up completely, allowing the presentation of the calculations as a handy table, so no calculations need be done at the time of topping up. It is important to note that the equation is an approximation due to slight volume changes on mixing, these are relatively small (c. 1–2% according to Waller and Strang (1996) and can be ignored for the purposes of this paper). This equation was used to produce Table 2 using Microsoft Excel© spreadsheet (Appendix 1). The desired concentration was arbitrarily set at 75%, but easily could be set differently. Concentrations to use for topping up have been approximated to the easily available concentrations, 96%, 88% (a 50:50 mix of 96 and 80%), 80%, and increments of 10% thereafter. Using larger increments is not recommended because this increases osmotic stress in topping up and the amount of rounding used in producing the table, making the process less accurate. A quick calculator also is provided for mixing topping-up solutions (Appendix 2).

An illustration is given below of how to find the concentration of alcohol with which to top up (Table 3). For example, to get the desired concentration of 75%, measure the concentration of alcohol initially in the jar and estimate the proportion of the jar filled with preservative (this can most quickly be done by eye, or with the aid of a calibrated dipstick). When calculating heights of fluid, a working head space is left at the top of the jar, usually 10 mm, so that the alcohol does not touch the seal (this is not included in height measurements; e.g., "100% full" would be the internal height of the jar minus 10 mm). Using Table 2, read across from the nearest concentration value on the left and read down from the nearest proportion value at the top. Where the readings cross is the concentration to use for topping up, e.g., for a jar that is 0.75 full of 67% alcohol, read across from the nearest value 67.5% and down from 0.75, which crosses at 96%. Top up with 96% to get a final concentration of around 75%.

Occasionally the readings cross in a square where there is no number. This either is because the alcohol is so concentrated that it cannot be brought to the desired concentration by the addition of water (small region to upper right of topping-up table) or, because the alcohol is so weak that it cannot be brought to the desired concentration by the addition of 96% alcohol (large region to lower right of topping-up table). In these cases it is still possible to top up successfully; however, some of the starting preservative must be discarded. The topping-up table can be used to show how much of the initial preservative must be discarded before topping up can proceed. For example, for a jar that is 0.75 full of 60% alcohol, read across from 60% and down from 0.75 proportion as above to find the square where the readings cross, then read back left to the next square containing a number (96% in the 0.60 proportion column). Pour away preservative until the proportion of the jar filled is 0.60 and then top up with 96%, for a final concentration or around 75%. An illustration of this is given in Table 4.

Testing the Topping-Up Table

The topping-up table was tested to confirm that it would work. Initially it was tested using a large calibrated measuring cylinder so that no specimens were put at risk, using

Table 2. Table for calculating the concentration of topping-up alcohol needed to return preservative concentration to 75% alcohol.

		Initial proportion of jar containing preservative																		
		0.05	0.10	0.15	0.20	0.25	0.30	0.35	0.40	0.45	0.50	0.55	0.60	0.65	0.70	0.75	0.80	0.85	0.90	0.95
Initial concentration of alcohol	100.0	70	70	70	70	70	60	60	60	50	50	40	40	30	20					
	97.5	70	70	70	70	70	70	60	60	60	50	50	40	30	20	10				
	95.0	70	70	70	70	70	70	60	60	60	60	50	50	40	30	20				
	92.5	70	70	70	70	70	70	70	60	60	60	50	50	40	30	20	10			
	90.0	70	70	70	70	70	70	70	70	60	60	60	50	50	40	30	20			
	87.5	70	70	70	70	70	70	70	70	60	60	60	60	50	50	40	30	0		
	85.0	70	70	70	70	70	70	70	70	70	70	60	60	60	50	50	40	20		
	82.5	70	70	70	70	70	70	70	70	70	70	70	60	60	60	50	50	30	10	
	80.0	70	70	70	70	70	70	70	70	70	70	70	70	70	60	60	60	50	30	
	77.5	70	70	70	70	70	70	70	70	70	70	70	70	70	70	70	70	60	50	30
	75.0	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80
	72.5	80	80	80	80	80	80	80	80	80	80	80	80	80	80	80	88	88	96	
	70.0	80	80	80	80	80	80	80	80	80	80	80	80	88	88	88	96			
	67.5	80	80	80	80	80	80	80	80	80	80	88	88	88	96	96				
	65.0	80	80	80	80	80	80	80	80	80	88	88	88	96	96					
	62.5	80	80	80	80	80	80	80	80	88	88	88	96	96						
	60.0	80	80	80	80	80	80	80	88	88	88	96	96							
	57.5	80	80	80	80	80	80	88	88	88	96	96								
	55.0	80	80	80	80	80	80	88	88	88	96	96								
	52.5	80	80	80	80	80	88	88	88	96	96									
	50.0	80	80	80	80	80	88	88	88	96	96									
	47.5	80	80	80	80	88	88	88	96	96										
	45.0	80	80	80	80	88	88	88	96	96										
	42.5	80	80	80	80	88	88	96	96											
	40.0	80	80	80	80	88	88	96	96											
	37.5	80	80	80	88	88	88	96	96											
	35.0	80	80	80	88	88	96	96												
	32.5	80	80	80	88	88	96	96												
	30.0	80	80	80	88	88	96	96												
	27.5	80	80	80	88	88	96													
	25.0	80	80	80	88	88	96													
	22.5	80	80	88	88	96	96													
	20.0	80	80	88	88	96	96													
	17.5	80	80	88	88	96	96													
	15.0	80	80	88	88	96														
	12.5	80	80	88	88	96														
	10.0	80	80	88	88	96														
	7.5	80	80	88	88	96														
	5.0	80	80	88	96	96														
	2.5	80	80	88	96	96														
	0.0	80	80	88	96	96														

alcohol solutions with a range of starting concentrations and volumes (all possible combinations of 0%, 20%, 40%, 60%, 80%, 96% and 0.1, 0.3, 0.6, 0.9 proportion of the cylinder containing preservative). These were topped up using the method outlined above. Once thoroughly mixed, concentration measurements were taken. The final concentrations were: mean 74.8%, range 71.1–78.2%, standard deviation $\pm 2.0\%$ ($n = 24$), giving a close and consistent approximation to the desired 75%.

Table 3. Illustration of how to find out the concentration of alcohol to top up with to get the desired concentration using Table 2.

		Proportion of jar filled				
		0.65	0.70	0.75	0.80	0.85
Initial concentration of alcohol	72.5	80	80	80	88	88
	70.0	88	88	88	96	
	67.5	88	96	96		
	65.0	96	96			
	62.5	96				

Next, the experiment was repeated with a selection of real jars containing specimens selected from the NHM entomology alcohol-preserved collection to represent a range of preservative volumes (0.05–1.00), concentrations (43.7–82.8%), jar types (old style ground glass, honey/jam jars, bail-top jars, etc.) and contents (large specimens, small specimens in vials, bulk samples). Specimens preserved in >85% were ignored because these mighty have been preserved for DNA studies. Specimens that had dried out were left dry, because they were stable and there was no need to rehydrate them at present. Because the jars were not calibrated, the proportion filled by preservative was estimated by eye. The type of jar and contents were recorded in case this had any effect. The final concentrations were: mean 75.7%, range 71.9–78.0%, standard deviation ±1.7% (n= 29), again giving a close approximation to the desired 75%. The observed errors are most likely due to: 1) the volume changes on mixing; 2) the rounding used to simplify the topping-up table; or 3) estimation of proportions by eye rather than by measuring. The slightly higher average overall final concentration probably is due to having specimens/vials, etc. that were mostly towards the bottom of the jars; this would cause the initial proportion of preservative in the jar to be slightly overestimated. In some jars with large amounts of cotton wadding (used to hold vials in place) mixing took some time; this caused spuriously high readings of concentration at least temporarily until the concentrated alcohol was mixed with the original weak alcohol held in the cotton wool. Thus, when checking these to see if the correct concentration had been reached in these instances, it was important to allow enough time for mixing.

In summary, the results from topping up real specimens show that the method is effective for topping up alcohol-preserved collections within an acceptable degree of

Table 4. Illustration of how to find out the concentration of alcohol with which to top up in situations where the alcohol is so concentrated that it cannot be brought to the desired concentration by the addition of water, or, where the alcohol is so weak that it cannot be brought to the desired concentration by the addition of 96% alcohol.

		Proportion of jar filled							
		0.50	0.55	0.60	0.65	0.70	0.75	0.80	0.85
Initial concentration of alcohol	65.0	88	88	88	96	96			
	62.5	88	88	96	96				
	60.0	88	96	96	←	←	←		
	57.5	96	96						

accuracy (±3%). Therefore, there is every reason to think that the method is robust and will work for a wide variety of jar and specimen types.

The Effectiveness of Some Topping-Up Methods Over Time

Method

An experiment was conducted to evaluate the effectiveness of some different topping-up methods to maintain concentration at the arbitrary values 75% ±5%, compared with the new topping-up table. The evaporation of alcohol from specimen jars was simulated using 500-ml beakers. Evaporation was accelerated by leaving the beakers uncovered in order to get results within a reasonable time. Unlike the method used in Carter (1995), the amount of preservative remaining was measured by the proportion of the beaker filled with preservative, rather than measuring the mass by difference, so that the method was comparable to topping up of real jars where the mass of the jar could not be conveniently estimated. The experiment took place in the Natural History Museum's Darwin Centre entomology spirit store in the same conditions as the terrestrial invertebrate collection. The method was as follows:

- Six 500-ml beakers were each filled with 500 ml of 75% IMS and left uncovered.
- The jars were placed inside one of the collection cabinets in the store.
- For convenience, commonly available concentrations of alcohol were used for topping up, i.e., 80%, 96%, and 88% (a 50 : 50 mix of 80 and 90%).
- The beakers were inspected at approximately 4-day intervals, and if needed, topped up according to the following protocols:

    1. No topping up.
    2. Topping up with 80% when >10% volume lost.
    3. Topping up with 88% when >10% volume lost.
    4. Topping up with 96% when >10% volume lost.
    5. Topping up as follows: if concentration <60% and volume >50%, discard fluid until 50% left and top up with 96%; if concentration <60% and volume 33–50%, top up with 90%; if concentration <60% and volume 10–33%, top up with 80%. This method is based on a procedure described by van Dam (c. 2008) for maintaining specimens in aseptic range 50–80% using his plastic indicator beads. This differs, however, from the full method he used for anatomy collections (Van Dam, pers. comm.). This assumes no intervention because the specimen is exposed above the fluid, a reasonable assumption for most terrestrial invertebrate collections which are placed in small jars inside a larger jar, which prevents the specimens becoming exposed even at low alcohol volumes.
    6. Topping up using concentration shown by new topping-up table when >10% volume lost.

- Alcohol concentration and volume were measured at every inspection, and before and after topping up.
- When measuring alcohol concentration after topping up, the alcohol was thoroughly mixed.
- When measuring alcohol concentrations generally, the first reading was discarded to remove the risk of contamination from the previous sample in the measuring cell of the meter, and an average of three subsequent readings was taken.

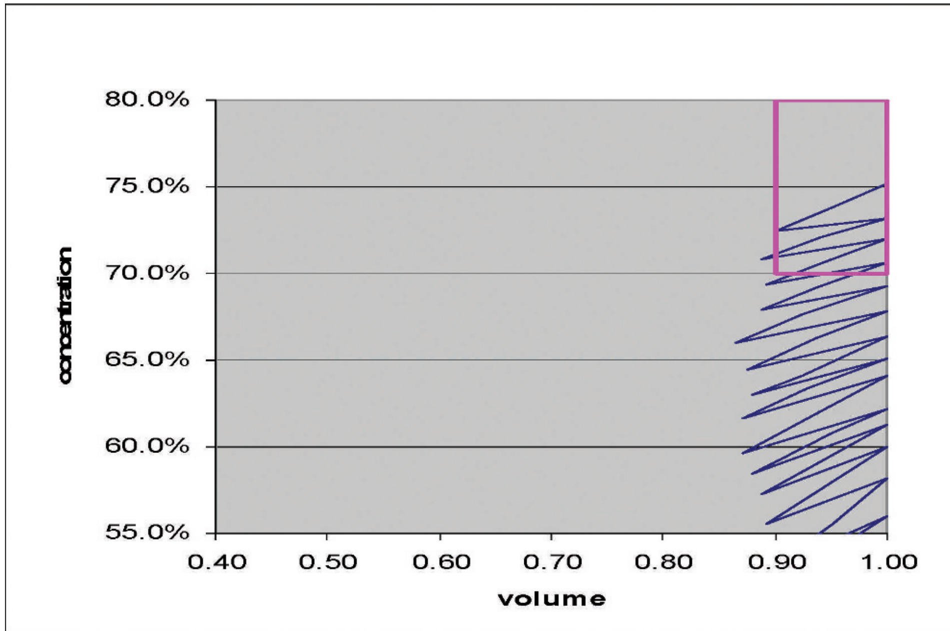

Figure 2. Combined concentration and volume changes over time in relation to ideal values (rectangle). Protocol 2: Topping up with 80% alcohol when >10% volume lost. This shows the so-called 80% fallacy, where continued use of 80% alcohol for topping up can lead to a steady reduction of alcohol concentration over time to very low levels.

- Temperature and the parts per million of alcohol in the air in the store was measured at each inspection to check the conditions were reasonably consistent.
- The environmental conditions in the store were measured to see if the evaporation rates predicted were correct and could be used as a broad indicator of what topping-up methods are likely to be needed.

Results

Protocol 1 (no topping up) showed a reduction both in volume and concentration; over time, the rate of decrease in concentration became slightly greater and the rate of decrease in volume became slightly less. The concentration went below 70% after 0.2 of the volume was lost. Protocols 2–4 showed a very similar pattern of volume changes with regular fluctuation between 0.9–1.0. Protocol 2 (topped up with 80% alcohol; Fig. 2) showed an overall trend of decreasing concentration, going below 70% after the loss of about 0.3 of the volume, reaching 60% after the loss of about one volume and dropping below 55% by the end of the experiment with no sign of levelling out. Protocol 3 (topped up with 88%) showed a similar, although less steep trend of decreasing concentration, going below 70% after the loss of about 0.5 of the volume, and below 65% by the end of the experiment, with no sign of levelling out. Protocol 4 (topped up with 96%) showed an overall trend of increasing concentration, barely exceeding 80% on occasion, appearing to level out just below 80%. Protocol 5 (Fig. 3) showed for each cycle of topping up, a steady reduction in concentration and volume down to a concentration of just below 60% and a volume of about 0.5. At this point the plastic bead floated, showing the concentration was just below 60%; the jar was topped up, causing a concentration change of about 20%.

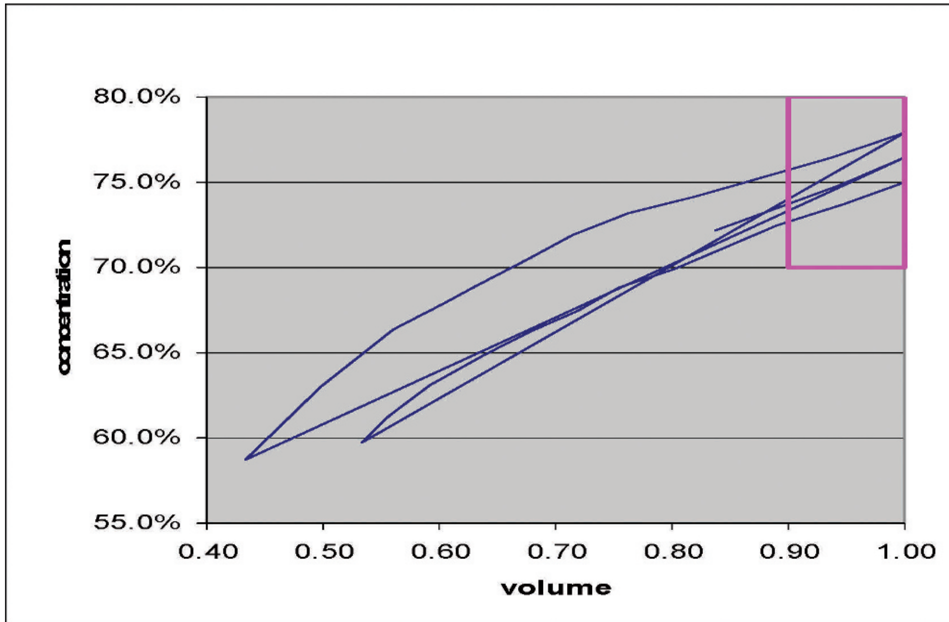

Figure 3. Combined concentration and volume changes over time in relation to ideal values (rectangle). Protocol 5: Topping up in order to maintain antiseptic range of concentration 50–80% as follows: if concentration <60% and volume >50%, discard fluid until 50% left and top up with 96%; if concentration <60% and volume 33–50%, top up with 90%; if concentration <60% and volume 10–33%, top up with 80%.

Protocol 6 (topped up according to the topping-up table; Fig. 4) showed oscillation of several percent around the desired concentration value of 75%. The environmental conditions in the store over the period monitored (Fig. 5) were somewhat variable within broad limits, with relative humidity at an average of 53.8% ± 8.5%.

Discussion

Although the experiment does not provide a realistic view of the timescale of evaporation, nor does it accurately model the permeation of alcohol/water vapour through a rubber seal, by comparison with empirical observations of collection profile and anecdotal observation of concentration changes in spirit jars, it does appear to show an approximate simulation of the related behaviour of concentration and volume for the differing topping-up methods in nonpermeable jars under the given conditions, and certainly resembles the situation of a broken seal. Some useful features can be noted: 1) the evaporation curve from Protocol 1 suggests topping up is needed at or before 0.1 of the volume is lost because this gives a safety margin before the concentration drops to 70% (after loss of 0.2 of the volume); 2) Protocol 2 gave similar results to those given by Carter (1995), that repeated topping up with 80% IMS is insufficient to maintain concentration; 3) similarly, for Protocol 3, 88% is insufficient to maintain concentration, although the rate of decrease is less; 4) Protocol 4 shows that 96% will cause the concentration to become slightly too concentrated; 5) the results for Protocol 5 show large concentration and volume changes and the potential for osmotic stress; in Protocol 5 the plastic beads were not needed to see that topping up was needed, given the obvious loss of volume; and 6) the results for Protocol 6 using the new topping-up table were the

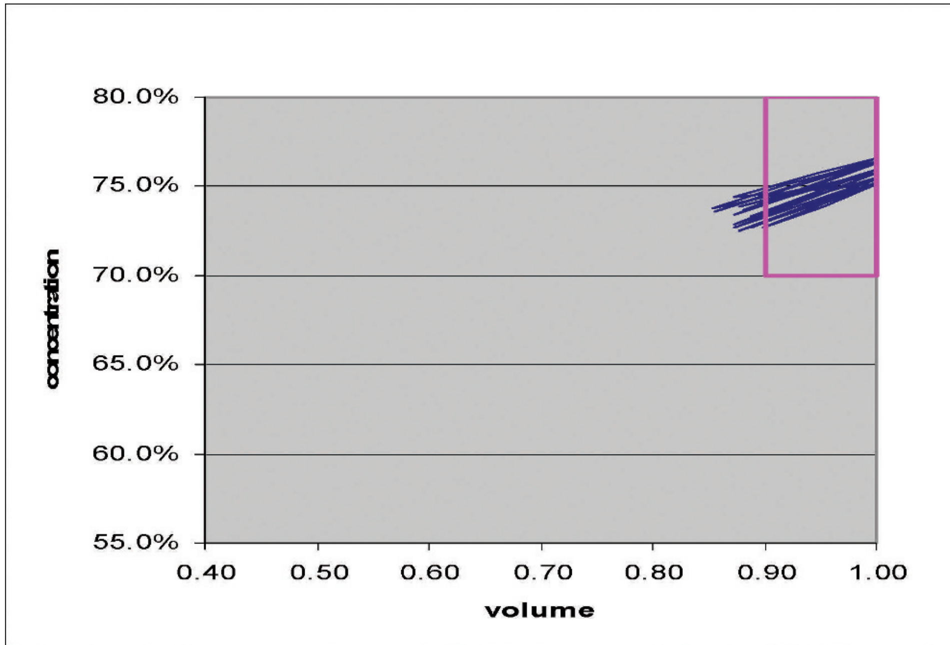

Figure 4. Combined concentration and volume changes over time in relation to ideal values (rectangle). Protocol 6: topping up using concentration shown by the new topping-up Table 2 when >10% volume lost.

best for maintaining concentration at a given level with a limited degree of fluctuation—in this case 75% ± 3%.

The success of the new topping-up table is shown by the combined concentration/volume plot (Fig. 4), which shows the graph almost entirely within the set limits (shown by the rectangle). This can be compared to the graphs for Protocols 2 and 5, which are mostly outside the set limits (Figs. 2, 3). The next best and simpler method was to top up using only 96%, although this became slightly over-concentrated and there was no guarantee that it would not become more so over longer time periods. Interpolation between the results for Protocols 3 and 4 show that topping-up alcohol of an intermediate concentration c. 93% would be approximately right for maintaining concentration under the conditions of the Darwin Centre store, although this approach could not be used on its own without monitoring and readjustment at intervals. However, because it would be quick to implement, it might be part of a method incorporating routine topping up with 93% and no monitoring most of the time, and topping up using monitoring and the new table at regular intervals (e.g., 3–4 years), so that any jars that had deviated when monitoring was not used could be corrected. The high relative humidity for the store (Fig. 5) also suggested that concentrated alcohol (>90% at least) should be used to top up, q.v. Waller and Strang (1996) who noted that 70% alcohol at 50% relative humidity loses vapour equivalent to 95% alcohol. The new topping-up method might be improved further if the calculations for the topping-up table could include the slight volume change on mixing between alcoholic solutions.

Because the new method has been designed with invertebrate collections at the NHM in mind, it might not be applicable equally to other kinds of alcohol-preserved collections. Assumptions underlying the method are not the same for all spirit collections,

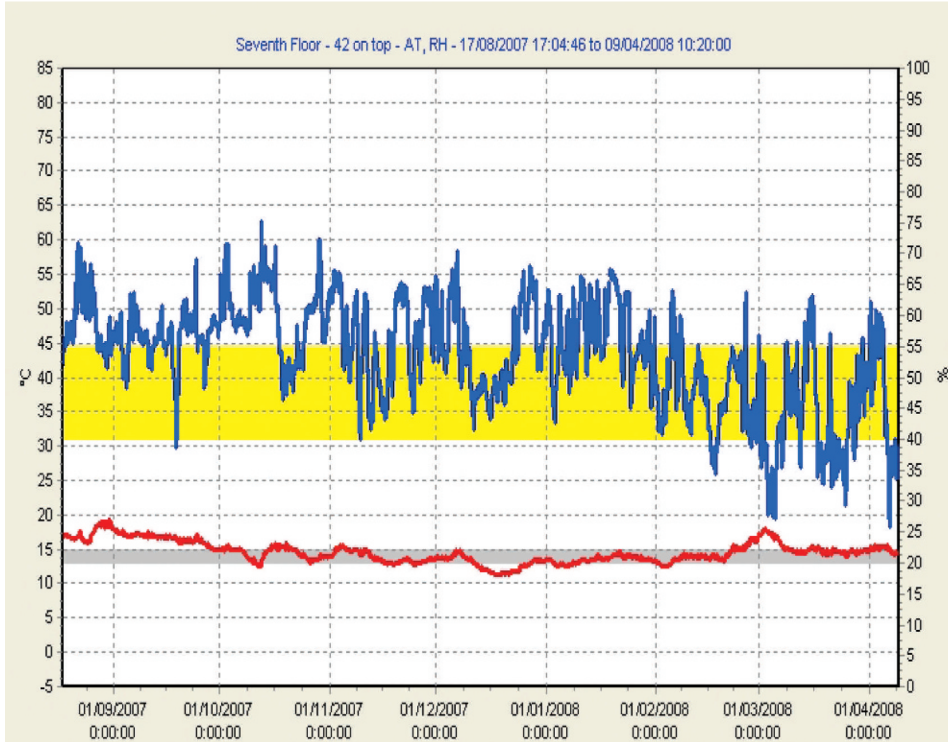

Figure 5. Environmental conditions in the Darwin Centre entomology spirit store. Upper trace and right hand scale is relative humidity; lower trace and left hand scale is temperature.

in particular that the collection is housed in standardised good-quality (Le Parfait-style) jars in a store with moderate to good environmental conditions. The new method (and any other topping-up method) will not compensate for poor storage containers or poor environmental conditions, which will require increased work in topping up. The method is slightly more time-consuming than some other methods (a few seconds per jar after initial set up) but this is traded off by the better quality of care of specimens. For large collections, if time for curation is limited, time can be better spent improving jars and environmental conditions, and staff temporarily can use a quicker but less-accurate topping-up method, such as topping up with c. 93% alcohol after every c. 10% volume loss as mentioned above the Darwin Centre store (n.b., this value depends on store conditions and might not be transferable to other stores), until conditions are stabilised. Recent recuration projects of sections of the entomology spirit collection at the Natural History Museum testing the new procedure have usefully combined remedial topping up with jar replacement without excessive time penalty.

In stores with fluctuating environmental conditions, leaks in some kind of jar seals might be caused if there is insufficient head space above the alcohol to allow for expansion of alcohol vapours (van Dam 2000). For example, rigid Bakelite lids can be cracked, ground glass jars can be loosened, and sealed jars can be cracked. This can be countered by using Le Parfait-style jars that have highly robust lids, where positive pressure on the seal is maintained by the spring wire top that can re-establish the seal after any loosening event. Even with Le Parfait-style jars, a small head space (c. 10 mm)

should be left so that the alcohol does not touch and degrade the seal, and so the jar does not overflow when it is opened. Where susceptible jar types have to be used, the new topping-up method should be practicable, providing that a larger head space is left to accommodate vapour expansion; the estimation of the initial and final heights of alcohol can be made to take account of this.

Occasionally the wrong solution is used when topping up, giving a concentration that is too weak. This can be prevented by: 1) measuring and labelling the concentration of stock solutions to be used; and 2) measuring the concentration in the jar after topping up. These actions take seconds per jar and should give a high degree of confidence. For those wishing a degree of redundancy, a plastic bead system might be used as a backup, although in line with the rest of the approach of this paper, to reduce fluctuations in concentration to a minimum, it would be better to use a bead that detects lowered concentration much sooner, for example, at 70% (if aiming at a target concentration of 75%). Only one bead per jar is needed to show that attention is needed. The human factor generally needs to be taken into account; most methods of topping up are time consuming to some extent, can be tedious, need some knowledge and equipment, and the environment of stores can be unpleasant. However there is little excuse for not devoting adequate time and resources, because such basic maintenance of the collection is a core part of the curator's job, and curators and managers need to prioritise and support these activities accordingly.

Developing a Practical Method for Topping Up

The following recommendations draw together the conclusions presented above on the properties of evaporating alcohol, the aims of topping up, general considerations gained from the collection profiling exercise, assessment of previous published methods, and the experiments on the effectiveness of topping-up regimes over time. The recommendations are made for use with terrestrial invertebrate collections, housed in good quality (Le Parfait-style) jars in a store with moderate to good environmental conditions. By way of a worked example, concentrations and volumes are shown for use with the Natural History Museum's terrestrial invertebrate spirit collection in the Darwin Centre store. It is important to note that, because of variation in the environmental conditions between stores, the same method might not work exactly elsewhere, but the same process can be followed to work out values appropriate for another collection and store.

Preliminary Considerations

Before undertaking any topping up:

- Undertake the health and safety risk assessments for using alcohol-based preservatives and obtain appropriate personal protective equipment.
- Decide the concentration of alcohol in which to store your particular specimens and the allowable range of fluctuation around this value. For the Darwin Centre store, this was set at 75 ± 5 volume % standardised to 20°C.
- Decide the volume at which to top up your specimens. This should be based on the normal rate of evaporation for the store (e.g., see the evaporation curve in Fig. 1), giving a margin for error before the concentration drops below the acceptable lower limit (second bullet point above), and giving a margin for error before any specimens are exposed. For the Darwin Centre store, this was set at 0.9 of the volume.
- Decide how often to inspect and top up if needed, and work out evaporation rates from different kinds of storage jars, both effective and defective. This should be based

on how long it takes the worst kind of jar to reduce the volume of preservative to the volume at which to top up (third bullet point above). For the Darwin Centre store, past experiences suggest annual losses in the region of c. 1% volume in jars with an effective seal and 5–10% in those with a defective seal, with some variation depending on jar and seal type—therefore, inspections must be done at least annually.

- Obtain a digital density meter that automatically converts readings to volume % standardised to 20°C, such as the Anton Paar DMA 35N or equivalent (Anton Paar 2000).
- Calculate a topping-up table similar to Table 2 based on the formula and method described above, and the desired concentration (second bulleted point above), print it out, preferably in colour, and seal it in a plastic pouch so it will be alcohol-resistant.
- If it is difficult to estimate proportions of the volume (e.g., some designs of jar that taper slightly) make a graduated dipstick for this kind of jar marked off in tenths.
- Obtain (or make up), verify, and clearly label the concentrations of the stock solutions of alcohol.
- The protocols below might need to be modified to allow a larger head space in cases where there is a risk of seal breakage from high vapour pressure (for susceptible jar types in stores with sudden temperature fluctuations).
- Ideally, protocols should include the monitoring of pH; however, measuring the pH of alcohol solutions is difficult and is best dealt with elsewhere.

Remedial Topping Up

This procedure is recommended, if starting with a neglected collection, or one at the wrong concentration:

- Make a profile of a proportion of the collection as described above, selecting systematically across the collection, to represent all parts of the collection; this should help estimate the amount of time and materials and any special problems.
- Check every jar.
- Check that the jar is not defective; if it is, replace it.
- Check that each jar is tall enough so that evaporation between inspections will not leave any specimen exposed; if this is likely, transfer the specimen to a larger jar.
- Check the concentration of alcohol and the proportion of the jar filled with preservative—do not assume full jars will be at the correct concentration.
- Calculate the concentration of the alcohol to add using the topping-up table.
- Fill the jar up with alcohol; jars should consistently be filled to the top (leaving c. 10 mm head space to avoid contact between alcohol and the seal). This allows subsequent visual detection of evaporation easier; if a large change of concentration (more than 5%) is needed, top up in stages to reduce osmotic stress
- Make a final check of the concentration, and adjust if needed.
- Make a record.
- If specimens are completely dried out, do not try to rehydrate them without good reason. They are usually stable when dry, and rehydration probably will cause more damage—leave a note in the jar saying "found dehydrated on such and such a date," and store at humidity and temperature levels appropriate for dried tissue samples (i.e., not necessarily in a spirit store).
- Jars with very high concentrations might have been preserved for DNA work; if so, clarify the purpose of preservation, and if they need to be kept at high concentration, label them clearly, and preferably transfer them to low temperature storage.

Routine Topping Up

This is recommended for collections that recently have undergone remedial topping up to the right concentration and volume. Every fourth or fifth time, a complete check is recommended as for remedial topping up:

- Set the timetable for inspection (fourth bullet point of preliminary consideration above) and stick to it.
- Top up all jars where volume is less than the volume decided above (third bullet point of preliminary consideration above).
- For each jar topped up, check to see if seal is defective and jar or seal need to be replaced.
- For specimens known or suspected to be at risk of acidification, replace the preservative completely.
- Check the concentration of the alcohol and the proportion of the jar filled with preservative.
- Calculate the concentration of the alcohol to add using the topping-up table (once the collection has been stabilised by remedial topping up; this should be straightforward because there should be relatively little variation in concentrations and volumes, and the table can be used to provide rules for common situations. For example, for the Darwin Centre store, if volume is reduced by about 10%: it is topped up with 96%, and if concentration $<73.75\%$, it is topped up with 88% if concentration $>73.75\%$).
- Fill up the jar with preservative; jars should consistently be filled to the top (leaving c. 10 mm head space to avoid contact between alcohol and the seal); this allows subsequent visual detection of evaporation easier.
- Check the topped-up jars, so the correct concentration is reached.

Do not underestimate the human factor—topping up can be tedious and stores are often cold, dull, and away from regular places of work. Persist to get topping up seen as a priority, and check that it has been done correctly. The use of clear and reasoned protocols should help staff appreciate the problems and implement improved collection care, because the method and the benefits will be seen clearly.

Additional Considerations

Making Life Easy

The following recommendations for managing alcohol-preserved collections should make the method easier:

- Over-concentrated solutions can be returned to a desired concentration gradually by adding 80%; this brings them down gradually over the course of several rounds of topping up, saves the need to mix up lower concentrations of topping-up alcohol, and reduces osmotic stress due to larger concentration changes.
- Improve store conditions: a reduction of temperature reduces evaporation rate; a reduction in humidity maintains the concentration of the remaining alcohol (Horie 1994).
- Set up a refrigeration unit (e.g., use a spark-proof freezer) for smaller insects that are susceptible to bleaching (e.g., Noyes 1990; Masner 1994), this also will reduce further evaporation from these jars, saving work on topping up.
- Replace defective or less effective jars.

- Standardise jars across the collection to one or a small range of sizes for ease of handling.
- Use Le Parfait-style glass jars. These are: 1) robust; 2) impermeable (except for the seal); 3) have quick release tops, for easy access; 4) have standard, easily replaceable seals; and 5) come in a range of convenient but standard sizes.
- Where consistent with access, use a smaller number of large jars where possible, because these are quicker to process than a larger number of small jars.
- Where consistent with access, use a smaller number of large jars because evaporation rates are relatively lower from large jars. Other things being equal, for an impermeable glass jar, for a larger jar of the same proportions, the volume of preservative contained increases as the cube of the linear dimensions, whereas the rate of evaporation should be proportional to the length of the seal, which only increases with the linear dimensions. Similarly for permeable jars the evaporation rate through the jar walls will be relatively less from larger jars because this should be proportional to the surface area of the jar, which increases only as the square of the linear dimensions.
- Specimens in small vials can be placed together in larger jars, so only one jar needs to be topped up instead of all the vials; vials also help buffer small specimens inside larger jars against concentration fluctuations.
- When using vials, take care to choose a good stopper (Levi 1966; Simmons 1995a); stoppers are often rubber, cork, metal screw caps with rubber inserts, cotton wool, and polyethylene. Rubber, cork, and most metals should be replaced because they quickly degrade in alcohol-based preservatives; cotton wool bungs allow alcohol to pass through and do not prevent the specimen from drying out if the jar dries out completely (although if stood upside down this will be put off as long as possible); polyethylene stoppers appear to be better in this respect, allowing very little interchange with fluid in the jar and give the best protection against drying out, although the behaviour of these bungs in alcohol needs to be monitored because their long-term stability is not proven.

In situations where: 1) a collection has undergone remedial topping up; 2) the store conditions are relatively stable over the long term, year on year; and 3) where a determination has been made of the concentration of alcohol which is likely to maintain concentration (page 17); it should be possible to top up routinely without monitoring each jar, although if this is done, full monitoring should be done every fourth or fifth time to check that topping up has been effective; effective record keeping is needed to remember when to monitor.

Further Considerations on the Retention or Disposal of Alcohol-Based Preservatives

Various considerations have been made of the benefits of retaining or disposing of preservatives, and concern the avoidance of leaching and the loss of leachates, the avoidance of acidification, and precipitates. Fluid preservatives act to some degree as solvents, and the fluid will contain components of the specimen that have been dissolved (Simmons 1995a). It has not been possible to trace any reference to the use of leachates for any purpose, so the value of leachates as a part of the specimen probably is not high; in any case many terrestrial invertebrates are collected by mass-sampling methods, so are associated with mixed leachates that cannot be separated or associated with the specimen of origin. In some cases, the leachates might be damaging to the specimen, particularly the acidic breakdown products of lipids. Further research is needed to determine the

incidence and effect of this process in terrestrial invertebrates; probably acidification is not common, although certain taxa, e.g., large larvae of some Diptera and Coleoptera, are particularly fatty and might be affected. Also, vegetable acids can leach from insect galls, e.g., tannic acid from cynipid galls on oak (*Quercus* spp.). Further research is needed to establish how acidification relates to risk to specimens, and to develop practical protocols for monitoring. Also, where lipid leaching is a problem, if the preservative is saturated with lipid and the alcohol concentration drops, leached lipids can precipitate out on the specimens (Simmons 1995a); this is another argument for limiting fluctuations in alcohol concentration. However, caution should be exercised before replacing discoloured preservatives because they sometimes reach equilibrium with the specimen and are not necessarily acidic or containing lipids. In such cases, if the preservative is replaced, leaching could resume and cause further degradation of the specimen.

Possible Areas for Further Research

The current paper suggests some areas of future research of value for improving the management of alcohol-preserved collections of terrestrial invertebrates. Firstly, the role of acidification needs to be clarified:

- What is the likely damage?
- What is the incidence and any taxa at particular risk?
- Do any methods help to mitigate the risk?
- Can an empirical risk-based estimate be made of the minimum tolerable values of pH, and estimates be made of the minimum time interval at which to check pH (Cato 1990)? and
- Is there a role for pH buffers and neutralising agents in alcohol-preserved insects as has been recommended for formaldehyde-preserved natural history specimens (Simmons 1991), and if so what might be their effect on specimens?

Secondly, the susceptibility of specimens to bleaching (Hancock 1985; Masner 1994) needs to be addressed:

- What is the likely process and how is this damage cause?
- Which taxa are most vulnerable?
- How can low temperature best be used to mitigate this effect?
- Are there any risks associated with the use of low temperature, such as the precipitation/crystallisation of solutes leading to tissue damage? and
- Is there any interaction between bleaching and acidification?

Thirdly:

- How significant is the buffering effect on concentration of vials inside larger jars? and
- Which is the best closure for vials inside larger jars, both in terms of buffering concentration and long-term stability?

Acknowledgments

To staff of the Natural History Museum, London: Howard Mendel, Claire Valentine, and Paul Brown for encouragement and checking the manuscript; Richard Sabin provided the graph of environmental conditions in the Darwin Centre spirit store and Clare Methold provided information on the procedures for disposal of waste alcohol; Andries van Dam and Simon Moore for a helpful review and discussion.

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Appendix 1. Method for calculating topping-up table using Microsoft Excel spreadsheet.

This method will work for any desired end concentration of alcohol:

1. Type the target concentration in cell a1.
2. Type in the table headings for the initial concentration of alcohol; these should be from "100" to "0" downwards from cell a2 (use the fill series tool for speed).
3. Type in the table headings, for the proportion of the jar initially filled with preservative; these should be from "0.05" to "0.95" rightwards from cell b1 (use the fill series tool for speed).
4. For the values in the body of the table, type the following formula in cell b2 "= (a1−$a2*b$1) / (1−b$1)" and copy the formula to all cells in the table by selecting the formula and dragging the fill handle.
5. Delete all values in the body of the table which are <0 and >100 because these are meaningless.
6. Round all values in the body of the table to the nearest concentration of topping-up alcohol you have available, e.g., 96%, 88%, 80%, 70%, 60%, 50%.
7. Make the table easy to read according to your preference, e.g., adjust the number of decimal places in the table headings to 1, and in the body of the table to 0; add table lines; embold headings; colour the background of the table cells to show the different concentrations of alcohol to use.
8. Print out in colour and laminate in an alcohol-resistant pouch.

Appendix 2. Quick calculator for making up topping-up solutions of different strengths, using commonly available concentrations of ethanol (80% and 96%). This table is approximate because of slight volume changes on mixing. Use only distilled or deionised water because tap water contains contaminants.

	Add the following proportions:		
Desired solution strength (%)	Distilled or deionised water	80%	96%
0	1.00		
10	0.88	0.12	
20	0.75	0.25	
30	0.62	0.38	
40	0.50	0.50	
50	0.38	0.62	
60	0.25	0.75	
70	0.12	0.88	
80		1.00	
88		0.50	0.50
96			1.00

DETECTING PESTICIDE RESIDUES ON MUSEUM OBJECTS IN CANADIAN COLLECTIONS—A SUMMARY OF SURVEYS SPANNING A TWENTY-YEAR PERIOD

P. Jane Sirois,[1] Jennifer Poulin,[1] and Tom Stone[2]

[1]*Analytical Research Laboratory, Canadian Conservation Institute, Department of Canadian Heritage, 1030 Innes Road, Ottawa, Ontario K1A 0M5, Canada*
[2]*Objects, Industrial Collections, Archaeology and Furniture Division, Canadian Conservation Institute, Department of Canadian Heritage, 1030 Innes Road, Ottawa, Ontario K1A 0M5, Canada*

Abstract.—The Canadian Conservation Institute (CCI) has provided analytical support to Canadian museums for over 20 years by assisting with the detection of pesticide residues in over 2,000 objects in various collections. In the 1980s primarily natural history collections were analyzed using x-ray fluorescence spectrometry (XRF) to detect arsenic and mercury. XRF was chosen because it could be performed on site, was nondestructive and produced no hazardous waste. The realization that ethnographic and anthropology collections also were potentially contaminated with pesticides led to testing objects in those collections as well. Requests to test educational collections to ensure that objects being handled by children were safe also were received. More recently, testing for organic pesticides has become part of on-site pesticide surveys. The pesticides tested for in most collection surveys are: arsenic-, mercury- and lead-based compounds, bromine residues, DDT (dichlorodiphenyltrichloroethane) and its analogues, and naphthalene and para-dichlorobenzene (PDB) in the storage atmosphere. When information in museum records suggests the use of other pesticides, testing would be undertaken to detect evidence of those compounds as well. The compiled survey data has provided information that has assisted museums to develop proper handling and storage guidelines for objects in their collections: undergoing conservation treatments, being sent out on loan and used in exhibitions, for research, and for other purposes.

Introduction

Accretions and white powders observed on museum objects can be a clue to the presence of pesticide residues. Not all white residues present on objects indicate hazardous pesticide residues, however. Some white powders such as sawdust, plaster, or cornstarch, can be residues from cleaning procedures or object preparation and might not be hazardous. Hazardous compounds such as arsenical compounds and other pesticides also might have been used during object preparation. The goal of the pesticide surveys undertaken by the Canadian Conservation Institute (CCI) is to detect pesticide residues in museum collections and individual objects so that museum staff has information on chemical compounds that might be possible hazards in their collections. An assessment of the potential health hazards involved is not the mandate or expertise of CCI. Once the hazardous objects are identified they then can be properly labeled and stored, thereby providing protection to the people working with them.

CCI's first pesticide survey, requested by a museum conservator, was performed in 1986 and included the analysis of both natural history specimens and some selected anthropology objects. Since then, over 20 surveys, both on-site surveys and the analysis of samples and wipes sent to CCI, have been undertaken. Museum objects such as those to be sent out on loan, objects that will be undergoing conservation treatment, First Nations objects to be repatriated (i.e., masks), and general anthropology and ethnographic collections now are being analyzed to detect pesticide residues. First Nations (Indian) and anthropology collections have been analyzed to detect the presence of arsenic, mercury, lead, and bromine, and occasionally specific organic pesticides.

Collection Forum 2010; 24(1–2):28–45

Requests for testing educational collections also have been received to ensure that museum objects potentially handled by children are safe.

Testing for organic pesticides is becoming part of our on-site pesticide surveys. Pesticides routinely screened for are: arsenic-, mercury- and lead-based compounds, bromine residues, DDT (dichlorodiphenyltrichloroethane) and related compounds, and naphthalene and para-dichlorobenzene (PDB) in the storage atmosphere. If information in museum records suggests the use of other pesticides, testing would be undertaken to try to detect evidence of those compounds as well.

History of Potential Pesticide Use in Canada

Literature research has provided some background information on the types of pesticides suggested for use in Canadian collections. Canadian museum publications from the early half of the 20th century suggest using many different pesticides to safeguard collections from insect infestation (Leechman 1931; Anderson 1948). In a 1929 Annual Report of the National Museums of Canada, a mixture of ethylene dichloride with one part carbon tetrachloride was suggested for use. Carbon disulfide, hydrocyanic acid gas, chloropicrin, naphthalene, and para-dichlorobenzene also were mentioned (Leechman 1931). Sodium fluoride was used to control cockroaches and a solution of mercuric chloride in alcohol was suggested as an effective fungicide (Leechman 1931). For synonyms and formulas of pesticides see Appendix 1.

Pesticides suggested for use in natural history collections in 1948 were: naphthalene, para-dichlorobenzene, arsenic–borax mixtures, arsenic–alum mixtures, arsenical soaps, DDT, sulfur, carbon disulfide, and a mixture of ethylene dichloride and carbon tetrachloride (Anderson 1948). White arsenic (arsenic trioxide) diluted in water or sodium arsenite diluted with water applied to skins to prevent infestation also was suggested (Anderson 1948). The recipe Anderson used for its preparation, based on an earlier 1925 recipe from Rowley was: ¼ pound (113 g) arsenic trioxide (white arsenic), ½ pound (227 g) carbonate of soda (washing soda), and 1 quart (946 ml) of water.

Another source of information on possible pesticides used was literature collected by the Canadian Museum of Civilization (CMC). These documents included correspondence on recommended methods of pest control from other museums sent to the National Research Council of Canada and then forwarded to what is now the CMC. For example, the use of carbon disulfide, naphthalene, and para-dichlorobenzene, and the treatment of specimens mounted for exhibition by the application of various solutions of arsenic compounds were recommended by the American Museum of Natural History (Anderson 1964). Dowfume, Paracide crystals, and arsenical soaps also were suggested for mothproofing in correspondence from the Smithsonian Institution (Hobbs 1964). Pamphlets on pest control distributed by Agriculture Canada in the 1960s and 70s mentioned the following pesticides: chlordane, DDT, diazinon, dieldrin, malathion, lindane, pyrethrum powder, and sodium fluorosilicate (Andison 1960; Creelman 1969; MacNay 1967a, 1967b). Mothproofing sprays available for household use around 1974 were reported to contain combinations of pesticides such as: methoxychlor with pyrethrum and piperonyl butoxide, or resmethrin with tetramethrin (Agriculture Canada 1974). Application of dusts containing 10% DDT, 5% chlordane, or 2% dieldrin were suggested by Agriculture Canada prior to laying of rugs in a 1965 pamphlet for the general public outlining the treatment of fabrics to prevent pest damage (MacNay 1965). It is not known whether these practices were adopted. This information does, however,

provide insight into methods and chemicals being recommended to the museum community, the general public, and other industries at the time.

For more current Canadian practices, CCI Technical Bulletin 15 (Dawson 1992) outlines chemical control methods used as a last resort for pest control when integrated pest management and nonchemical methods are ineffective. Chemicals indicated for pest eradication, not mentioned above, are: propoxur, chlorpyrifos, silica aerogel, bendiocarb, carbaryl, methyl bromide, ethylene oxide, sulfuryl fluoride, pentachlorophenol, zinc naphthenate, phosphine, and diatomaceous earth. At the time of writing, these pesticides all still were available in at least one registered product in Canada (Health Canada 2010). The Pest Management Regulatory Agency of Health Canada oversees the regulation of pesticide use and registers products for use in Canada through the Pest Control Products Act and Regulations (Bulletin 15, p. 1). Some provinces, regions, or municipalities can further ban federally approved products.

As the potential use of an increasing number of pesticides on museum objects became apparent, screening for more organic pesticides was included in our site surveys when there was documentation of use (e.g., DDT use was vaguely documented in some museums). The advancement of analytical instrumentation also has led to lower detection limits, more efficient analysis, and the ability to analyze a wider range of compounds.

Nineteen on-site surveys have been undertaken since 1988 (1,758 objects). Other analyses also have been undertaken to detect pesticide residues; however, these analyses either have been done on objects sent to CCI or on samples sent to the laboratory.

Methods of Analysis

Nondestructive X-ray Fluorescence Spectrometry (XRF)

X-ray fluorescence spectrometry was used primarily for the nondestructive detection of arsenic, mercury, and lead in on-site surveys. The objects analyzed in these studies were selected by museum staff to obtain a representative selection of objects from their collection. No samples were taken for those analyses. XRF is predominantly a surface technique that indicates the presence of arsenic, mercury, lead, bromine, and generally elements above atomic number 13 (aluminium) in the periodic table. It might not accurately indicate the amount present in the object, however. For example, the presence of a thick layer of fur or feathers between the contaminated area and the detector can result in the underestimation of the amount of arsenic present in the skin.

Since 1986, the equipment and experimental parameters used for XRF analysis have changed. In 1986 a Tracor Northern NS-570 x-ray energy spectrometer equipped with an I-125 radioisotope source and a lithium-drifted silicon detector was used. Two areas per object were analyzed for a time of 200–300 seconds/area. The lower limit of detection for arsenic using this technique was determined experimentally to be comparable to a 500 ppm (0.05%) reference material of arsenic, prepared as arsenic trioxide in Chemplex™ x-ray mix. The reference materials were made by combining Chemplex x-ray mix with arsenic trioxide or mercuric sulfide (HgS) and preparing them as briquettes.

From 1995 to 2004 a Canberra Packard "Inspector" portable x-ray energy spectrometer equipped with a lithium-drifted silicon x-ray detector and a Cd-109 radioisotope source was used. Between one and three areas were analyzed on each large object for a count time of 200 seconds/area. Only one area was examined on small objects. A series of reference materials for arsenic and mercury was analyzed to determine the lower limits of detection for these elements and to determine the approximate corresponding x-ray peak areas at different concentrations. The lower limit of detection

Table 1. Lower limits of detection of the Innov-X Systems handheld XRF spectrometer.

Soil mode		Filter mode	
Element	Unit (ppm)	Element	Unit (μm/cm^2)
As	8	As	6
Pb	15	Pb	0.4
Hg	20		

for arsenic again was determined to be comparable to a 500 ppm reference material. The reference materials used were the same Chemplex x-ray mix-based arsenic trioxide or mercuric sulfide (HgS) briquettes mentioned above.

Starting in November 2004, an Innov-X Systems handheld XRF spectrometer with an x-ray tube source was used for the analysis. The objects were analyzed using a layer of polyethylene between the spectrometer and the object to prevent cross-contamination and to avoid direct contact of the metal face of the spectrometer with the object. A minimum of two areas were examined on each large or composite object using a count time of 60 seconds per area. Replicate analyses were performed on several objects to ensure that variability was due to the artifact itself and not the spectrometer. The lower limits of detection determined are listed in Table 1. The Chemplex x-ray mix-based briquette reference materials were used to determine the accuracy of the instrument in soil mode for grouping the results into the categories outlined in Table 2 (used to analyze thick samples). NIST lead paint standards on mylar sheets, SRMs 2579a, were used for lead in filter or thin film mode.

Thin areas of the objects, such as feathers, were analyzed using the filter mode that reports the results in μg/cm^2, whereas thicker areas were analyzed using the soil mode, which reports the results in ppm.

The XRF results categories are presented in Table 2.

Analysis Using Samples

Several types of analysis have been performed by CCI's Analytical Research Laboratory using samples. The types of samples taken for the various techniques used are outlined below.

Wipe Samples.—Wipe samples have been taken for analysis by atomic absorption spectrophotometry (AAS), inductively coupled plasma spectrometry (ICP), or XRF.

Table 2. XRF results categories.

Pre-November 2004[a]		Post-November 2004	
ND[b]	<500 ppm	ND	
		NQ[c]	
Trace	500 pmm to <0.1%	Trace	25–100 ppm
		Low	100–1000 ppm or <0.1%
Minor/Moderate[d]	0.1% to <1%	Moderate	0.1% to <1%
High	1–5%	High	1–5%
Very high	>5%	Very high	>5%

[a] November 2004 marks the date hand-held XRF was introduced.
[b] ND (not detected): below the lower limit of detection for the specific element. See Table 1.
[c] NQ (not quantifiable): three times the detection limit.
[d] Objects with readings in the minor/moderate classifications and higher are considered contaminated and should be handled with caution.

Moist towelettes, supplied by the lab contracted to perform the ICP analysis, are used to sample the object surface for the determination of arsenic, mercury, and lead. Wipe samples for XRF analysis have been prepared by rubbing a Johnson and Johnson First Aid® brand gauze square moistened with distilled water in an "S" stroke as described in the literature over a 10×10 cm area selected on the object using a template (Makos 2001; McDermott 2004). These wipes are then enclosed in Mylar-lined cardboard coin holders prior to analysis.

Powder and Object Samples.—When powders were seen on the surface of the object, the storage container, or object mount, scrapings were taken using a scalpel and the samples were brought back to the laboratory for subsequent analysis. Small samples of the object itself, generally being less than 2 mm^2 in size, also were taken sometimes for analysis. These types of samples (powders, fibers, skin, and feathers) were used for x-ray diffraction (XRD), polarized light microscopy (PLM), scanning electron microscopy/energy dispersive x-ray spectrometry (SEM/EDS), gas chromatography–mass spectrometry (GC–MS), and Fourier transform infrared spectrometry (FTIR) analysis.

Swab Samples.—Cotton swabs were used to sample objects by lightly drawing the swab(s) across surfaces and into crevices where unseen residues might have collected. Two different sample collection methods have been used, one using a template in which the swab was used in a manner similar to the S stroke method mentioned earlier, and a less systematic way where the surface and crevices were rubbed. The less systematic way provided qualitative results only, but rubbing in the crevices better detects pesticide residues possibly still remaining from early applications. Prior to making the cotton-tipped swabs, the cotton batting was precleaned in an acetone solvent bath. These samples are typically used for GC–MS.

Microvacuum Filters.—Microvacuum samples were collected using a low-flow, hand-held vacuum pump which draws particles from the surface of the objects and traps them onto individual glass filters contained inside preloaded, 37 mm, clear, 3-piece, plastic cartridges. A piece of Tygon® tubing, approximately 1.5 inches (4 cm) long, sliced diagonally on the end in contact with the object, formed the vacuum nozzle. This was attached to the plastic cartridge for sampling. The artifacts were sampled using one of the methods described earlier for approximately 30 seconds. These nozzles were used only once before being discarded, ensuring that there was no cross contamination. The cartridges then were sealed with the caps provided and stored at room temperature for future extraction and analysis by GC–MS.

Passive diffusion air cartridges.—Passive diffusion air cartridges, left to collect and store volatiles such as dichlorvos and naphthalene in the air of a storage facility for a set period of time (typically overnight), have been used to provide information on some contaminants present in the storage environment.

Sample Preparation and Analytical Techniques

Wipe Samples for ICP or XRF.—Wipe samples for ICP analysis generally have been provided by the analytical laboratory undertaking the analysis. The wipe, once used, was enclosed in the container provided and then sent to the lab. To prepare wipe samples for XRF, the gauze pads (wipes) were folded into an approximately 1.5- by 1.25- inch (3.8- by 3.2-cm) rectangle and placed into a Mylar-lined, cardboard coin flip or a plastic Ziploc bag for XRF analysis.

Scanning Electron Microscopy/Energy Dispersive X-Ray Spectrometry (SEM/EDS).—Samples for SEM/EDS can be powders, skin, feathers, or fibers and are

Table 3. Reporting levels for DDT, DDE, DDD, perthane, and methoxychlor.

	Levels (μg)	
Category	DDT, DDE, DDD, perthane	Methoxychlor
ND[a]		
NQ[b]		
Trace	0–0.1	0–1
Low	0.1–5	1–50
Moderate	5–10	50–100
High	10–30	100–300

[a] ND (not detected): below the lower limit of detection for the specific element.
[b] NQ (not quantifiable): three times the detection limit.

prepared by mounting them on carbon planchets using double-sided carbon tape, then carbon coating them. SEM/EDS of the samples reported here was performed using a Hitachi S-530 SEM integrated with a lithium-drifted silicon x-ray detector and a Noran Voyager x-ray microanalysis system. Using this technique, elemental analysis of volumes down to a few cubic micrometers can be obtained for elements from sodium (Na) to uranium (U) depending on the detector. In a study done in 1989, the lower limit of detection for arsenic was determined experimentally to be 0.2% (Sirois and Taylor 1989) and 0.1% in 2000 (Sirois and Sansoucy 2001). In one museum survey, a comparison of the results of samples analyzed by two techniques, SEM/EDS and spot tests, was undertaken. The SEM/EDS analyses agreed with the ‘‘Weber’’ arsenic spot test results (Hawks and Williams 1986), thereby confirming the usefulness of these spot tests (see Found and Helwig 1995 for details on the methods of analysis used).

Gas Chromatography–Mass Spectrometry (GC–MS).—The sampling methods used most often for the analysis of organic pesticides were: microvacuum filtering, cotton swabs, and room air sampling with passive diffusion air cartridges. The samples of particulate matter removed from the objects via swabbing or through microvacuuming were extracted in the appropriate solvent (acetone) overnight, left to dry, and the residue was redissolved in a small quantity of solvent to concentrate pesticides that might be present. Acetone solubilised the organic pesticides of interest, including: organophosphates (e.g., dichlorvos, diazinon, malathion); organochlorines (e.g., DDT, dichlorodiphenyldichloroethane [DDD], dichlorodiphenyldichloroethylene [DDE], methoxychlor, dichlorobenzene); and carbamates (fenobucarb, terbucarb) (Murayama et al. 2000; Schmidt 2001).

Unused cotton swabs and glass filters also were extracted and analyzed as previously described to serve as ‘‘blanks.’’

The results from the analysis of the organic pesticide samples were reported as listed in Table 3.

Passive diffusion air cartridges also were analyzed by GC–MS. The charcoal membranes from the cartridges were extracted in an appropriate organic solvent prior to analysis by GC–MS.

Results

Site Surveys

Objects originally were examined only for arsenic and mercury. The range of pesticide residues investigated has broadened to include: arsenic, mercury, lead, bromine residues, dichlorvos, naphthalene, PDB, methoxychlor, DDT, perthane, DDD, and DDE. Prior to

Table 4. Percentage of positive XRF results for As, Hg, Pb, Br, and organic pesticides for results up to 2000 (after Table 1 in Collection Forum, Sirois 2001).

			% of objects that tested positive					
Museum and object type	Date	No. objects	As	As > 1%	Hg	Pb	Br	Organic pesticides
Museum A	1986–1987							
Total		110	68	19	8		22	Not tested
Ornithology		43	81	26	2		19	
Mammalogy		41	78	29	12		10	
Anthropology		24	33	—	17		46	
Other		2	50	—	—		—	
Museum B	1995							
Total		173	67	32	5		8	Not tested
Ornithology		130	81	40	4		2	
Mammalogy		43	25	7	1		23	
Museum C	1996							
Total		130	95	42	4		1.5	Not tested
Ornithology		124	91	41	4		1.5	
Mammalogy		6	67	67	—		—	
Museum D, Ornithology	1999	114	98	29	3		—	Not tested
Museum E	1999							
Total		155	73	11	5		6	Not tested
Ornithology		144	74	12	6		5	
Other		11	55	—	—		18	
Museum F, Anthropology	1999	111	9	—	1		19	Not tested
Museum G, Anthropology	2000	71	42	—	18	65	86	Not tested
Museum H, Anthropology; Masks	1999	6	33 (trace)	—	33 (paint)		—	Not tested
Museum I, Anthropology; Masks	1999	8	13 (trace)	—	—		—	Not tested

the site surveys, we requested that the museum provide us with information on the potential pesticides that might have been used so that we could prepare for the appropriate testing and the approximate number and type of objects to be analyzed. A list of the specific objects to be analyzed is not generally provided prior to our arriving on site.

The percentages of positive readings of objects selected for analysis from various Canadian museum collections are listed in Tables 4, 5, and 6. It should be noted for both the XRF analysis and SEM/EDS analysis that different areas of the same object often contain very different concentrations of a particular compound (Found and Helwig 1995), thus affecting the results. This is more important with minute samples than when performing XRF on a larger area of the object.

Table 4 is a modified version of Table 1 from Sirois 2001 and is included to provide a comprehensive look at contaminants detected in Canadian collections. The surveys reported are broken down by museum and year and the objects analyzed are categorized by collection type to help establish trends in the different collections. Table 5 includes results from surveys since 2001. The data in Tables 4 and 5 were reported separately (i.e., not combined into one table) because the results were reported in different ways.

Table 5. Percentage of positive As, Hg, Pb, and Br XRF analyses and organic pesticides for results from 2001–2007 (including wipe samples).

Museum and object type	Date	No. objects	% of objects that tested positive					Organic pesticides
			As	As > 1%	Hg	Pb	Br	
Museum J, Anthropology	2001	16	13	—	19	100	—	Not tested
Museum K, Anthropology	2001	224	75	1	15	71	49	Tested bagged air of 8 objects for dichlorvos, naphthalene, and para-dichlorobenzene (PDB). Naphthalene and PDB detected in 7/8 objects. Dichlorvos not detected.
Museum L	2003							
Total		62	87	10	2	—	—	Not tested
Ornithology		38	95	11	3	—	—	
Mammalogy		16	81	6	—	—	—	
Other		8	63	13	—	—	—	
Museum M	2003							Methoxychlor / DDT / Perthane
Anthropology		89	26	—	4	58	10	43% / 26% / 29%
1 natural history					NQ[a]			
Museum N	2003							
Total		105	9	1	3	16	12	Not tested
Anthropology		101	8	—	3	17	13	
Natural History		4	25	25	—	—	—	
Museum O	2003							Air quality tests were done by the provincial ministry of labor for para-dichlorobenzene, naphthalene, and arsenic. All were well within the legal limits.
Total		126	32	9	3	8		
Education		19	10	—	—	10	37	
Ornithology		70	54	14	13	16	49	
Mammalogy		25	36	—	4	—	52	
Anthropology		12	8	—	—	67	42	

Table 5. Continued.

Museum and object type	Date	No. objects	% of objects that tested positive					Organic pesticides				
			As	As > 1%	Hg	Pb	Br	Methoxychlor	DDT	DDE	DDD	Perthane
Museum P	2004											
Total		59	83	—	39	83	5	71%	37%	19%	19%	14%
Anthropology[a]		37	76	—	24	84	3	70%	22%	11%	11%	—
Chilkat[a]		22	95	—	77	82	9	73%	64%	32%	32%	36%
Museum Q	2005											
Education[a]		140	11	1	2	21	26	Not tested				
Wipes		17	—	—	—	—	N/A[b]					
Museum R	2005							Not tested				
Dioramas[a] Total		37	86	8	27	84	N/A					
Ornithology[a]		15	87	20	60	67	N/A					
Mammalogy[a]		22	86	—	5	95	N/A					
Museum S	2006											
Wipes; fur collection		22	—	—	—	—	N/A					

[a] Includes NQ (not quantifiable) as positive.
[b] N/A = not analysed/not reported.

Table 6. Percentage of positive SEM/EDS results for As, Hg, Pb, Br, and organic pesticides for results to 2007[a].

Museum and object type	Date	No. objects	% of objects that tested positive					Possible organic pesticides[b]
			As	As > 1%	Hg	Pb	Br	
Museum T	1993							
Total		61	52		3			Not tested
Ornithology		37	76		0			
Mammalogy		24	17		8			
Museum U	2000							Not tested
Swabs; total		143	28		1			By SEM/EDS chlorine was detected in 85% of samples; phosphorous in 14%.
Ornithology		112	32		—			
Mammalogy		31	7		3			
Museum V	2001	83	2		—	—	—	Not tested
Samples; total								Cl detected in most samples
Ornithology		24	4		—	—	—	
Mammalogy		58	3		—	—	—	
Other		1	—		—	—	—	

[a] Including data from Table 2, in Collection Forum, Sirois 2001.
[b] The chlorine detected could be due to chlorinated organic pesticides or possibly to salt (NaCl) used in the preparation of specimens.

Table 6 summarizes the SEM/EDS results of samples from museum objects. The results of these two techniques, XRF and SEM/EDS, are not directly comparable; however, both are indicators of whether or not contaminants are present.

As is noted at the bottom of both tables, results in the NQ category were considered as positive. These values are, however, in the realm of what would be considered a naturally occurring level of arsenic in soil (1–40 ppm arsenic) the average value being 3–4 ppm (ASTDR 2007).

The results from these analyses were intended only to advise on the presence or absence of mercury and/or arsenic and their relative values in the objects examined and not to provide an evaluation of the health risk. An assessment of the potential health hazards involved is not the mandate or expertise of CCI. Because the goal of pesticide surveys is to identify chemical compounds that might pose hazards in the collections, the representative selection of objects by museum staff for analysis might be inclined more towards those objects suspected of being contaminated. As a consequence, the survey results might be slightly skewed in the positive direction. The reports are intended to assist the museum and appropriate authorities in assessing the problem and in establishing proper procedures.

Samples

Powder samples collected during site surveys were analyzed to identify the compounds on the objects. XRD, FTIR, PLM and GC–MS were the techniques used to identify the powder samples. Compounds identified to date, both hazardous and not, are:

Table 7. Occurrence of elements identified in collections (by XRF or sample analysis).

	As		Hg		Br
Collection	XRF	Samples	XRF	Samples	Samples
Ornithology (%)	84	38	6	0	8
Mammalogy (%)	56	7	5	3	18
Anthropology (%)	41	0	12	0	32
Education (%)	11	N/A	2	N/A	27
Other (%)	29	N/A	0	N/A	10

- arsenical powders and compounds such as arsenic trioxide
- lead arsenate
- mercuric chloride
- organic pesticides: DDT, DDD, and DDE, methoxychlor, naphthalene, para-dichlorobenzene, lindane, perthane, and terbutol
- alum, borax, salt, cornstarch, Epsom salts, calcite, kaolin, and titanium dioxide

Discussion

Inorganic Pesticides

When comparing the data from different surveys, the changes in method and data collected must be factored in. Early on we were looking primarily for the presence of arsenic and mercury, with lead and bromine being secondary considerations. Different methods were used as well, depending on the particular contaminant being investigated or whether a site survey was possible. Sometimes samples were sent to CCI because it was not possible to arrange a site visit within the required time frame.

The collections studied were: ornithology, mammalogy, anthropology, education, and ‘‘other,’’ which includes fish, reptiles, and mollusks. Because the methods and elements examined were similar for all these collections studied, overall averages were computed for arsenic and mercury.

Arsenic most frequently was detected in ornithology collections when analyzed using XRF (see Table 7), followed by mammalogy collections, and lastly anthropology and education collections. Different collections seem to have different pesticide profiles, an example being a collection of Chilkat blankets from Museum P where low levels of mercury were found in over 75% of the objects examined. It also generally was observed that the percentage of objects that tested positive for arsenic was much higher when the analysis was carried out using XRF on the whole specimen than when samples were analyzed by SEM/EDS. Different collections were surveyed with these two methods, so it is not possible to compare the results directly. It might, however, be due to the arsenic present in the specimen not being present in detectable amounts on the exterior feathers or fur sampled, but being present inside the skin. The lower detection limit of the XRF technique also might contribute to this.

The difference in XRF results and the SEM/EDS results of samples for arsenic and mercury could indicate that negative values are obtained in samples, when in fact the object might contain arsenic or mercury. This could argue against using samples only to determine if arsenic or mercury is present. It shows that arsenic is not present on the outer surface but does not provide information on the object as a whole. Earlier XRF analyses of the exterior surface of mammal skins indicated high concentrations of arsenic in the

Table 8. Occurrences of organic pesticides identified in two anthropology collections.

Chemical	Museum M	Museum P	Average
Number of objects	89	59	
Methoxychlor (%)	43	71	54
DDT (%)	26	37	30
Perthane (%)	29	14	22
DDD (%)	3	19	7
DDE (%)	ND[a]	19	7

[a] ND = not detected.

areas analyzed and showed only minor amounts of arsenic present when samples were examined by SEM/EDS or with spot tests. Gloves wiped over a highly contaminated specimen also tested negatively for arsenic by SEM/EDS (Sirois and Taylor 1989).

Organic Pesticides

The data from organic pesticide analysis also revealed that different collections have specific problems. For example, chlorine (Cl) was detected in most of the samples from Museum V and Museum U; organic pesticides were detected in two anthropology collections from Museum M and Museum P. These four examples highlight the individual characteristics of each collection.

Pesticide survey results for DDT and related chlorinated organic pesticides performed on two anthropology collections are shown in Table 8. An average value also is listed. Other pesticide residues detected in a few objects (five or fewer) in the collection of Museum M were: DEHP = di(ethylhexyl)phthalate (5), terbutol (1), nicotine (2), and chlorophenyl isocyanate (1).

Naphthalene or para-dichlorobenzene were sampled in three collections using organic vapor canisters and subsequently analyzed by GC–MS to determine whether these chemicals were present (Museum K, Museum M, and Museum O). Neither of these compounds was detected with organic vapor cartridges at Museum M; however the compounds were detected in the other two museums but at levels below the allowable limits.

Use of Results

The results for each object are provided to the museum conservation and/or curatorial staff to indicate which objects in their collection contain pesticide residues. If pesticide residues are detected in an object, it is labeled with the contaminant identified and the semiquantitative value (i.e., high arsenic, see Table 2) before the object is returned to storage. It is the responsibility of the museum to seek assistance from an occupational or industrial hygienist, or the provincial health and safely authority, to ensure safe procedures for handling are adopted. If requested, information on certified industrial hygienists and contact information for the provincial health and safety authorities is provided.

Conclusions

Testing objects in collections has many benefits. It has clarified the issue of whether "a white powder" observed by a conservator is potentially harmful or not in specific cases. It also has indicated that collections attributed to specific museums or collectors might have been subjected to certain pesticide regimes. This type of information is useful in

predicting possible residues in related but unanalyzed objects. The statistics obtained from the various types of Canadian collections indicate the likelihood of finding arsenic is greatest in ornithology collections, followed by mammalogy, anthropology, and education collections. The presence of other organic pesticides in two collections has been confirmed.

XRF testing quickly, easily, and nondestructively can detect inorganic elements present in most objects made of organic materials to alert the user to contamination. Further analysis of samples will more definitively determine whether the contaminant is on the interior or exterior of the object.

Health and safety protocols and handling guidelines have been developed in conjunction with Health Canada for CCI staff dealing with potentially contaminated objects in museum collections to ensure staff safety because pesticide surveys can involve handling and moving up to 200 potentially contaminated objects in a period of 2–3 days.

Acknowledgments

The authors would like to acknowledge and thank the many people who have contributed to this body of work over the years. It is truly a collaborative effort and without each and every person and institution, we would not have the body of knowledge available to us today that we do. Thanks to: Kate Helwig, Tom Strang, Marie-Claude Corbeil, Ian N. M. Wainwright, John Taylor, Elizabeth Moffatt, CCI; Martha Segal, CMC; Lisa Bengston, Royal BC Museum; Mima Kapches, Ken Lister, Julia Fenn, Ronnie Burbank, Royal Ontario Museum; Anne MacKay, McCord Museum; David M. Green, Pamela Costanzo, Redpath Museum; Kasey Brewer, Royal Saskatchewan Museum and Royal BC Museum; Heather Dumka, Glenbow Museum; Margot Brunn, Royal Alberta Museum; Diane Komejan, Luci Cipera, Canadian Museum of Nature; Dave Benson, Chatham-Kent Museum; Serge Gauthier, Musée du Séminaire de Sherbrooke/Musée de la nature et des sciences; Carol Brynjolfson, Museum of Vancouver; Wayne Wood, Joseph Vincelli, Robert Rob, McGill University Environmental Safety Office; Kaleb Grittner, Saskatchewan Advanced Education Employment and Labour, Occupational Health and Safety; Fraser Fort George Museum; Luc Maheux and Lisa Bazzul, Workplace Health and Public Safety Programme, Health Canada.

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Appendix 1. Chemical formulas and names of compounds suggested for combating pest problems in Canadian collections.

Pesticide/Chemical	Chemical names	Synonyms	CAS #	Merck #	Formula
Alum	Aluminum potassium sulfate dodecahydrate[1]		10043-67-1	360	$KAl(SO_4)_2 \cdot 12H_2O$
Arsenic trioxide	Arsenic trioxide	White arsenic; arsenous acid; arsenous oxide; arsenous sesquioxide; arsenous acid anhydride[1,4]	1327-53-3	804	As_2O_3
Bendiocarb	2,2-Dimethyl-1,3-benzodioxol-4-ol methylcarbamate	Ficam; NC-6897[1]	22781-23-3	1036	$C_{11}H_{13}NO_4$
Borax	Sodium tetraborate decahydrate	Disodium tetraborate decahydrate; borax decahydrate; boric acid; sodium borate; boracic acid; borsaure; orthoboric acid[1,2,3,4]	1303-96-4	8590	$Na_2B_4O_7 \cdot 10H_2O$
Carbaryl	1-Naphthalenol methylcarbamate	Methyl carbamic acid 1-naphthyl ester; 1-naphthyl N-methylcarbamate; Sevin; ENT-23969; OMS-29; UC-7744; Carylderm; Derbac; Ravyon[1]	63-25-2	1787	$C_{12}H_{11}NO_2$
Carbon disulfide	Carbon disulfide	Carbon bisulfide; dithiocarbonic anhydride[1]	75-15-0	1811	CS_2
Carbon tetrachloride	Carbon tetrachloride	Tetrachloromethane; perchloromethane[1]	56-23-5	1816	CCl_4
Chlordane	1,2,4,5,6,7,8,8-Octachloro-2,3,3a,4,7,7a-hexahydro-4,7-methano-1*H*-indene[1,3]	Chlordan; Velsicol 1068; CD-68; Toxichlor; Octa-Klor; Octachlor; Ortho-Klor; Synklor; Niran; Corodane	57-74-9	2082	$C_{10}H_6Cl_8$
Chloropicrin	Trichloronitromethane	Nitrochloroform; acquinite; Larvacide 100; Picfume[1,3]	76-06-2	2156	CCl_3NO_2
Chlorpyrifos	Phosphorothioic acid *O,O*-diethyl *O*-(3,5,6-trichloro-2-pyridinyl) ester	*O,O*-diethyl *O*-3,5,6-trichloro-2-pyridyl phosphorothioate; chlorpyrifos-ethyl; phosphorothioate; Dursban; Lorsban; Dowco 179; ENT 27311; Affront; Empire; Lock-On; Pyrinex[1,4]	2921-88-2	2189	$C_9H_{11}Cl_3NO_3PS$
Diatomaceous earth		Infusorial earth; siliceous earth; fossil flour; kieselguhr; Celite; Super-Cel[4]			
Diazinon	Phosphorothioic acid *O,O*-diethyl *O*-[6-methyl-2-(1-methylethyl)-4-pyrimidinyl]ester	Basudin; Dazzel; Gardentox; Kayazol; Knox Out; Nucidol; Spectracide; Diazol; Diazitol; Parasitex; Sarolex; Neocidol; Antigal; G-24480; dimpylate; ENT 19507[1,4,5]	333-41-5	2995	$C_{12}H_{21}N_2O_3PS$

Appendix 1. Continued.

Pesticide/Chemical	Chemical names	Synonyms	CAS #	Merck #	Formula
DDT	1,1′-(2,2,2-Trichloroethylidene)-bis[4-chlorobenzene]	Dichlorodiphenyltrichloroethane; chlorophenothane; clofenotane; dicophane; pentachlorin; Gesapon; Gesarex; Gesarol[1,3]	50-29-3	2841	$C_{14}H_9Cl_5$
Dichlorvos	Phosphoric acid 2,2-dichloro-ethenyl dimethyl ester	Phosphoric acid 2,2-dichlorovinyl dimethyl ester; dichlorophos; dichlorfos; dichlorovos; DDVP; SD-1750; Atgard; Dedevap; Divipan; Doom; Nuvan; Task; Vapona[1,4]	62-73-7	3079	$C_4H_7Cl_2O_4P$
Dieldrin	(1aα, 2β, 2aα, 3β, 6β, 6aα, 7β, 7aα)-3,4, 5,6,9,9- Hexachloro-1a,2,2a,3,6,6a,7,7a-octahydro-2,7:3,6-dimethanonaphth[2,3-b]oxirene	Compound 497; Octalox; HEOD; ENT-16225; Insecticide No. 497[1,3]	60-57-1	3103	$C_{12}H_8Cl_6O$
Ethylene dichloride	1,2-Dichloroethane	Dichloroethane; ethylene chloride; EDC; Dowfume 75; Dutch liquid; Brocide[1,3,6]	107-06-2	3797	$C_2H_4Cl_2$
Ethylene oxide	1,2-Epoxyethane	Dimethylene oxide; Carboxide; Oxyfume 12; Oxirane; Penngas; Epoxyethane; ETO; Anprolene [1,4,7]	75-21-8	3802	C_2H_4O
Hydrocyanic acid gas	Hydrogen cyanide	Prussic acid; formonitrile	74-90-8	4795	HCN
Lindane	(1α,2α,3β,4α,5α,6β)-1,2,3,4,5,6-Hexachlorocyclohexane	γ-HCH; γ-benzene hexachloride; gamma-benzene hexachloride; γ-BHC; gamma hexachlor; Aphtiria; Gamma-Col; Gammexane; ENT-7796; Jacutin; Lintox; Lorexane; Quellada; Kwell; Forlin; Gamophex; Scabecid[1,3,4]	58-89-9	5501	$C_6H_6Cl_6$
Malathion	[(Dimethoxyphosphinothioyl)thio] butanedioic acid diethyl ester	Malathon; carbophos; carbofos; Cythion 4049; Mercaptothion; phosphothion; insecticide no. 4049; ENT 17034; Derbac-M; Malamar 50; Prioderm; Suleo-M Malaspray; Malathiazol; Malathiazoo; Organoderm[1,3,4]	121-75-5	5700	$C_{10}H_{19}O_6PS_2$
Mercuric chloride	Mercury (II) chloride	Corrosive sublimate; mercury bichloride; mercury perchloride; corrosive mercury chloride[3]	7487-94-7	5876	$HgCl_2$

Appendix 1. Continued.

Pesticide/Chemical	Chemical names	Synonyms	CAS #	Merck #	Formula
Methoxychlor	1,1′-(2,2,2-Trichloroethylindene)bis[4-methoxybenzene]	Methoxy-DDT; Marlate; Prentox; Methoxcide; DMDT [1,3]	72-43-5	5990	$C_{16}H_{15}Cl_3O_2$
Methyl bromide	Bromomethane	Monobromomethane; Embafume; Meth-O-Gas; Terr-O-Gas; Maltox[1,4]	74-83-9	6029	CH_3Br
Naphthalene		Naphthalin; tar camphor; white tar; naphthene, moth flakes, mothballs, moth crystals[1,3,4]	91-20-3	6370	$C_{10}H_8$
Para-dichlorobenzene	1-4-Dichlorobenzene	*p*-dichlorobenzene; p-chlorophenyl chloride; Paracide; Paradow; Para-Di; p-DCB, 1,4-DCB; PDB; PDCB; PARA; Para-zene; Di-chloricide; Paramoth[1,3,4]	106-46-7	3057	$C_6H_4Cl_2$
Pentachlorophenol	2,3,4,5,6-Pentachlorophenol	Penta; PCP; penchlorol; Santophen 20[1]	87-86-5	7109	C_6HCl_5O
Phosphine	Hydrogen phosphide	Phostoxin; Celphos; Delicia Gastoxin; Detio-Gas-Ex-T; Ex-B[1,4]	7803-51-2	7338	H_3P
Piperonyl butoxide	5-[[2-(2-Butoxyethoxy)ethoxy]methyl]-6-propyl-1,3-benzodioxole[1]	Butylcarbityl (6-propylpiperonyl) ether; synthetic pyrethroid; Butacide; Pybuthrin; ENT-14250[1,6,8]	51-03-6	7476	$C_{19}H_{30}O_5$
Propoxur	2-(1-Methylethoxy)phenol methylcarbamate;	*O*-Isopropoxyphenyl *N*-methylcarbamate; aprocarb; Bay 39007; Bay 9010; Baygon; Bifex; Blattanex; Propyon; Suncide; Unden[1]	114-26-1	7836	$C_{11}H_{15}NO_3$
Pyrethrum powder	Active insecticidal constituents of pyrethrum flowers[1]	Pyrethrins; natural pyrethrins; Pyrethrum I or II; Cinerin I or II; Jasmolin I or II[3]		7964	$C_{21}H_{28}O_3$ I: $C_{22}H_{28}O_5$ II: $C_{20}H_{28}O_3$
Resmethrin	2,2-Dimethyl-3-(2-methyl-1-propenyl)cyclopropanecarboxylic acid [5-(phenylmethyl)-3-furanyl]methyl ester	Synthetic pyrethroid; Chryson; Crossfire; Derringer; FMC-17370; Isathrine; NRDC 104; Pynosect; Raid Flying Insect Killer; Respond; Scourge; Sun-Bugger #4; Synthrin; Syntox; Vectrin; Whitmire PT-110; SBP-1382[1,9]	10453-86-8	8153	$C_{22}H_{26}O_3$
Silca aerogel	Precipitated silica	Silicic acid; silicon dioxide; amorphous hydrated silica; silica xerogel; silica gel; Dri-die 67[1,4]	1343-98-2	8490	H_2SiO_3 (Approximately)
Sodium arsenite	Sodium meta-arsenite[1]		7784-46-5	8580	$AsHNaO_2$

Appendix 1. Continued.

Pesticide/Chemical	Chemical names	Synonyms	CAS #	Merck #	Formula
Sodium carbonate	Sodium carbonate	Carbonate of soda; washing soda; soda ash; Solvay Soda[1]	497-19-8	8596	Na_2CO_3
Sodium fluoride	Sodium fluoride	Chemifluor; Duraphat; Florocid; Fluoros; Flura-Drops; Karidim; Lemoflur; Luride-SF; Ossalin; Ossin; Osteo-F; Osteofluor; Slow-Fluoride; Villiaumite; Zymafluor; Flouridine; sodium monofluoride[1,7]	7681-49-4	8618	NaF
Sodium fluosilicate	Sodium hexafluorosilicate	Sodium silicofluoride; disodium hexafluorosilicate; sodium fluorosilicate; Salufer[1]	16893-85-9	8624	$Na_2Si\ F_6$
Sulfur			7704-34-9	8969	S
Sulphuryl fluoride	Sulphuryl fluoride	Vikane; sulfuryl fluoride; sulphuric oxyfluoride;Profume[1,4]	2699-79-8	8981	SO_2F_2
Tetramethrin	2,2-Dimethyl-3-(2-methyl-1-propenyl)cyclopropanecarboxylic acid (1,3,4,5,6,7-hexahydro-1,3-dioxo-2H-isoindol-2-yl)methyl ester	Synthetic pyrethroid; phthalthrin; FMC-9260; SP-1103; Neo-Pynamin[1]	7696-12-0	9223	$C_{19}H_{25}NO_4$
Zinc naphthenate	Zinc naphthenate	Naphthalic acid zinc salt	12001-85-3		$2(C_{11}H_7O_2)^{\cdot}Zn$

Note: The *Chemical Name* mentioned in the center column of this table is the Chemical Abstracts Name, if present, as it appears in the Merck index, 14th edition. For other chemical names see the Merck Index and other sources: 1) O'Neil 2006; 2) U.S. Borax, Inc 2008; 3) Hathaway and Proctor 2004; 4) Dawson 1992; 5) Extoxnet 1996a; 6) Odegaard and Sadongei 2005; 7) NIOSH 2005; 8) Pesticide Action Network Website, Piperonyl Butoxide, 2008; and 9) Extoxnet 1996b.

A CURATORIAL ASSESSMENT FOR STRATIGRAPHIC COLLECTIONS TO DETERMINE SUITABILITY FOR INCORPORATION INTO A SYSTEMATIC COLLECTION

Susan H. Butts,[1] Jessica A. Bazeley,[1] and Derek E. G. Briggs[1,2]

[1]*Division of Invertebrate Paleontology, Peabody Museum of Natural History, Yale University, 170 Whitney Avenue, PO Box 208118, New Haven, CT 065220-8118*
[2]*Department of Geology and Geophysics, Yale University, 210 Whitney Avenue, PO Box 208109, New Haven, CT 06520-8109*

Abstract.—The Schuchert Collection of the Yale Peabody Museum, Division of Invertebrate Paleontology (YPM-IP) Schuchert Brachiopod Collection is one of the largest brachiopod collections in North America and is the most heavily used collection in the division. A recently completed project involved the incorporation of large portions of the YPM-IP stratigraphic collection into our systematic collection, including cataloging, georeferencing, photography, and improvement of long-term storage conditions. A significant development in this project was the creation of a curatorial assessment to inventory and prioritize those components of the collection better suited for incorporation into the division's systematic collection and to create a hierarchy for removal of this component.

The following factors were considered: presence/absence of specimen data (locality or accession), bulk content (graded on a continuum with 1.0 representing individual fossils free from matrix and 0 representing bulk rock), percentage brachiopods, percentage of specimens with taxonomic determinations, and percentage with specific locality information. The output of the survey rates individual drawers with scores ranging from 0 to 100: 100 represents drawers that were ideal candidates for incorporation into the systematic collection, and 0 represents material more suited to a stratigraphic arrangement. Although the equation was developed specifically for the particular needs of this project and the collection strengths of the YPM-IP division, it is easily customized for a wide range of cross-disciplinary and highly specific collection applications.

Introduction

The Yale Peabody Museum, Division of Invertebrate Paleontology (YPM-IP) has two major brachiopod collections, the systematic collection (widely known as the "Schuchert Collection," because the nucleus is material collected or acquired by brachiopod specialist Charles Schuchert and his graduate students and colleagues) and the stratigraphic collection. The primary objective of this project was to relocate the inaccessible stratigraphic collection which is of tremendous scientific value, but resides in an environment of considerable risk. Encompassed in this goal particularly was to make the brachiopods scientifically useful to future researchers and to increase their educational value to students and the nonscience community. Large portions of the stratigraphic collection were incorporated into the systematic collection, and concurrent with the relocation of materials, the specimens were upgraded to the highest curatorial standards and an online, publicly available, object record (specimen or specimen lot) and image database was created for brachiopods and all other taxa relocated with the brachiopods.

The stratigraphic collection includes 3,858 drawers of material collected and acquired by IP curators as well as material collected by doctoral research students of Yale University. A portion of the collection includes bulk rock specimens (fossils with lithological remnants), which are essential for answering many questions in paleoecology and taphonomy. Paleontological research based upon collections increasingly extends beyond taxonomy and phylogeny to consider aspects of the paleobiology of organisms

Collection Forum 2010; 24(1–2):46–51

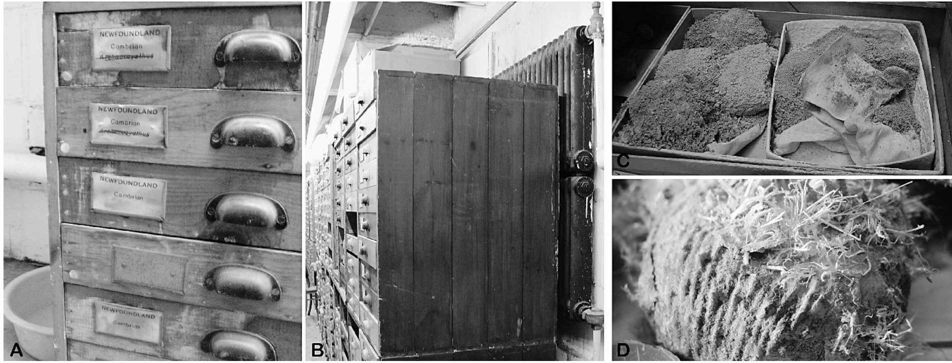

Figure 1. Conditions in the stratigraphic collection, Peabody Museum basement at the beginning of the project. A. Water-damaged drawers with drip bucket; B. Oak cabinets adjacent to steam heater; C. *In situ* acid erosion of carbonate specimens; D. Salt deposits (calcium sulfate) on a specimen, an indication of Byne's efflorescence.

and their relationship with other taxa and the environment (paleoecology). Bulk samples provide data on the matrix lithology that can be analyzed with modern methods (e.g., scanning electron microscope, electron microprobe, cathodoluminescence) to extract information on ocean and atmospheric geochemistry; the relationship of host lithology to types of preservation; the substrate preference of organisms, populations, and communities; and global climate change through time. A preliminary online digital *inventory* (not database) existed prior to this project for the stratigraphic collection with percentage of taxa (phylum or class) in each drawer and brief locality and stratigraphic information.

The Stratigraphic Collection

The historical age and state of the stratigraphic collection created special needs that required immediate attention to insure proper conservation for future use. It was at significant risk where it was housed in the basement of the Peabody Museum building, a structure built in the 1920s. Several agents of deterioration (Waller 1994) were present, including: water (flood risk); extreme fluctuations in temperature and relative humidity; contamination; physical forces resulting from overcrowded conditions and improper containers; data dissociation via blanket and abbreviated labeling practices; and loss of data from disintegrating and dirty labels. The YPM basement was subject to persistent leaks through the foundation, resulting in moisture damage, such as the warping of drawers and the discoloration of labels (Fig. 1A). The YPM basement is not climate-controlled and fluctuations of nearly 30°F and 15% relative humidity are observed routinely. Warm and humid conditions have resulted in the chemical erosion of trays and their velvet tray linings. Most of the cabinets in the stratigraphic collection were made in the mid 1900s of oak, which is highly acidic (Fig. 1B). The remaining drawers are newer but are constructed from plywood, the resins from which also create acidic conditions. In extreme cases, acidic conditions have caused *in situ* chemical erosion of specimens (Figs. 1C, 1D). Labels and other paper products bore signs of pest infestation, such as "scalloped edges" and faint grooves in paper surfaces. Specimens were overcrowded and most were in shallow nonacid-free trays, which allowed specimens to "migrate" from the original tray to an adjacent one during opening or transport, resulting in misleading association of data.

$$\Sigma = pb\,\frac{(2r + \tfrac{1}{2}s)}{(1.025 - l)}$$

p specimen data (locality or accession)
1 present
0 absent
b bulk content
1 individual specimens - 0 bulk samples
r % brachiopods (approx.)
s % taxonomically determined (approx.)
l % with specific locality information.
Σ (ranges between 0 and 100)
100 ideal candidate for incorporation into Systematic Collection for NSF-BRC
0 remains in Stratigraphic Collection

Figure 2. Curatorial assessment equation.

Many specimens collected in the mid- and late 1800s remained in the original collection boxes. Open storage had resulted in thick dust and soot deposition across specimens, labels, and trays. Several generations of specimen labels also were present in most trays. Some consisted of ripped notebook sheets scripted with fountain pen. In a few cases, ink had faded to the point where there was danger of complete loss of data. Thick dust made some labels difficult to read. Drawers frequently contained "blanket labels" that applied to the entire drawer contents (e.g., stating only the origin and accession number). If specimens were relocated to a different drawer, objects could become dissociated from data. Taxon data were often abbreviated.

Despite poor storage conditions, many elements of the stratigraphic collection met high curation standards for specimen identification, documentation of collection locality, and collection party information, and accession history—specimens potentially useful to researchers if they were organized systematically. An *informal* assessment of targets for incorporation of specimens indicated that approximately 45% of the stratigraphic collection could be incorporated into the systematic collection without encountering problems due to lack of taxonomic determinations or locality data. Because of the enormous volume of material, it was necessary to create a more robust tool to assess the collection in order to prioritize the transfer of material from the stratigraphic to the systematic collection.

Curatorial Assessment Equation

A curatorial assessment equation (Fig. 2) was developed to analyze quantitatively the suitability of individual drawers to be housed in a systematic versus a stratigraphic arrangement. Variables considered in the equation are: gross specimen data availability (accession and locality), bulk rock content, brachiopod abundance, taxonomic specificity, and specific locality data (coordinates or detailed written geographic description, collector name and date, and stratigraphic details).

Specimen data.—Specimen data (p) include any coarse-scale information associated with an individual drawer, such as a person's name, locality data, or an accession

number. A drawer with no information was assigned the value "0," whereas a drawer with information of any kind was assigned the value "1." If a drawer of specimens has no distinguishing characteristics to link it to any person, place, or collection, then it is of little, if any, scientific value. By including this variable in the equation, any drawer lacking basic information is automatically assigned the lowest possible score (Σ value). This variable is used as a multiplier to ensure that any drawer that lacks information, regardless of presence of taxonomically determined specimens, is disqualified from incorporation into the systematic collection.

Bulk content versus individual specimens.—This variable (b) assesses whether a particular drawer contains individual specimens, generally loose from the matrix, that would be well-suited to incorporation into the systematic collection. Because the contents are estimated visually, only five values were considered for this variable: 0, 0.25, 0.5, 0.75 and 1. A value of "0" indicates that the drawer contains exclusively bulk material, and is therefore unsuitable for incorporation into the systematic collection. Conversely, a value of "1" indicates that the drawer is made up entirely of individual specimens that have already been sorted (to some extent) taxonomically, and that the drawer should be incorporated into the systematic collection. The other values indicate intermediate proportions of individual specimens versus bulk material within a single drawer. By using this variable as multiplier, a drawer consisting solely of bulk material is excluded from consideration.

r = *% Brachiopods*.—A priority in the movement of collections was to incorporate brachiopods, because the Schuchert Brachiopod Collection at the YPM is the most frequently utilized part of our divisional holdings. Although all specimens in a drawer would be relocated from the stratigraphic collection to the systematic collection, we could preferentially incorporate brachiopod-rich drawers into our systematic collection by introducing this variable. The value r records the percentage of individual brachiopod specimens in each drawer. In the equation, the variables r, l, and s are expressed as a decimal values, rather than as a percentages, so that 100% = 1.0, 50% = 0.5, and 10% = 0.1. If a drawer contains rock slabs, the percentage of surface area of that slab occupied by brachiopods versus other organisms was estimated. Thus this variable estimates the brachiopods relative to the total number of specimens (or space occupied by specimens) within a drawer. The degree to which the drawer is filled is not relevant. Thus, the actual number of specimens was not taken into account, but rather the relative spatial area of brachiopod versus nonbrachiopod material. Two brachiopods in a small tray carry the same weight in this variable as two hundred brachiopods in a small tray. The value of r is doubled in the equation to reflect the importance of brachiopods in this assessment.

s = *% Identified*.—This value is an estimate of the decimal value (1.0 = 100%) of specimens that have taxonomic determinations. Many collectors identified their specimens prior to placing them in our collections. Other specimens have been identified since their collection date by curators, collection managers, museum staff, graduate students, and visiting scientists. This variable represents any name placed on a specimen, including common names. Many specimens are identified to the species level, but this variable also accepts "sponge" or "trilobite" as a name. This variable does not take into account the validity or accuracy of the taxonomic determination; it merely records that a name has been given to the specimen. As with the previous variable, the amount reflects spatial area occupied rather than number of specimens. The value of s is halved in the equation because identifications can be made at a future date. Incorporation is easier if the specimen has been identified, but there is value in unidentified specimens.

l = % *Locality*.—The locality variable accounts for any locality information: geographic, stratigraphic, or chronologic. Labels in the stratigraphic collection might include only a date and an accession number, but these two pieces of information can be related to data in field notebooks and used to plot an exact locality. Other labels might include only a country name or perhaps a geologic period. However, all locality information has been weighted evenly for this assessment. For a collection of this size, it was unreasonable to identify the quality of the locality data of the specimens in each drawer. As in the case of the previous two variables, the decimal value of specimens with locality information was treated as a percentage of spatial area within a drawer, and not by number of localities or number of specimens. This value is treated as a decimal percentage in the equation. The value of l is subtracted from 1.025 to ensure a nonzero denominator. This variable was weighted more heavily than the identification variable. Identifications can be made at any time, but locality data, once lost, are not easily retrieved.

Other factors considered during the evaluation.—Because every drawer was opened and visually inspected, we were afforded an excellent opportunity to assess the curatorial grade of the collection using the "Curatorial Continuum" method outlined in Hughes et al. (2000). The lowest level of this continuum (grade 1) denotes specimens that were simply acquired by the institution and accessioned. The highest level of this continuum (grade 5) represents specimens with locality data which have been identified and sorted taxonomically, are fully prepared and trayed, have been cataloged and databased, and are accessible to the scientific community. The intermediate grades represent various levels of curatorial care ranging from specimens with locality information through specimens that have been cataloged for dissemination through a database. Included in the assessment was fossil condition as a means of recording damage such as Byne's disease (mineral efflorescence), pyrite decay, and pest damage. We also noted other variables of all material in the drawers that might affect the rate at which material can be processed during transfer, such as presence of thin sections and other preparations, wrapping such as bags or other material, and fragile or mounted specimens.

Results

A total of 3,858 drawers were analyzed using our curatorial assessment. Drawers received scores ranging from 0 to 100, where a score of 100 represents drawers that were ideal candidates for incorporation into the systematic collection (individual specimens, mostly brachiopods, possessing taxonomic determinations and locality information). We found that 4.1% (158 drawers) of the collection were a perfect match for our criteria for incorporation into the systematic collection; 18.1% (700 drawers) received a score over 40, which was considered the minimum score for incorporation into the systematic collection; and 24.2% (934 drawers) received the lowest score obtainable, which means that they are suited only to storage in a stratigraphic arrangement. Over the course of 3.5 years, 20.9% of drawers (805 drawers) were incorporated into the systematic collection. This is greater than our predicted value based on our "perfect criteria" threshold. Beyond this threshold, specimens had lower percentages of brachiopods (therefore not "ideal" for the goal of our grant), but were still suitable for incorporation into the systematic collection. The specimens in these drawers have been retrayed in archival acid-free trays, labels have been cleaned with Groomstick natural rubber surface dry-cleaner, and, in the case of fragile or deteriorated labels, Mylar sleeves have been constructed to provide additional protection. Nearly 115,000 new specimen lots (nearly

70% brachiopods), representing over 270,000 individuals, have been cataloged in the YPM KE (Knowledge Enterprises) EMu database system, and over 45,000 object records have been modified. These are mirrored to a web-accessible searchable collections database. Concurrent with the project to incorporate portions of the stratigraphic collection into the systematic collection, a photographic initiative focusing on brachiopod specimens which were relocated into the systematic collection resulted in the addition of nearly 47,000 images of 23,250 specimens to the same online database (accomplished through rapid imaging practices using voice recognition software—see Butts et al. 2008). Damage (pyrite oxidation, Byne's disease, and/or pest damage) was reported in 85 drawers and these have been recommended for conservation.

Conclusions

Development and use of a curatorial assessment equation facilitated the identification of material in the stratigraphic collection most suitable for incorporation into the systematic collection. The analysis, requiring 3 months of skilled full-time employee labor, was a considerable investment in resources; however, it prevented wasted time during cataloging and incorporating materials into the systematic collection, because taxonomic determinations and researching and creating new localities are extremely time consuming (and require persons with advanced training in geology or biology). As the drawers with the lower curatorial assessment scores were being incorporated into the systematic collection, the per-drawer processing rate increased significantly and we could not have processed so many specimens in a 3-year span if drawers had not been prioritized using the curatorial assessment equation.

The curatorial assessment equation outlined above was designed specifically for the needs of the YPM-IP and tailored to address our need to relocate brachiopod specimens from the stratigraphic collection to the systematic collection. However, by changing the coefficients used to weight certain variables (taking into account the rationale we have provided, which can either be mathematical or subjective), the equation can be customized to handle a broad range of applications in museum paleontological collections.

Acknowledgments

This work was funded by the National Science Foundation, Biological Research Collections (NSF-BRC Grant Award #0545210—Conserving and incorporating the stratigraphic collection of Brachiopods into the systematic collection [the "Schuchert Collection"] and development of an online database). This manuscript benefited from reviews by Paul Mayer, Janet Waddington, and an anonymous reviewer.

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METAL-LID JARS IN MUSEUM COLLECTIONS?

ANDERS WARÉN, EMILY DOCK-ÅKERMAN, AND HANNA TAYLOR

Department of Invertebrates, Swedish Museum of Natural History, Box 50007, SE-10405 Stockholm, Sweden (anders.waren@nrm.se; emily.dock@nrm.se; hanna.taylor@nrm.se)

Abstract.—We report experiences from 25 years' use of glass jars with metal twist-on lids intended for food preservation. We have quantified the evaporation from the jars and simulated corrosion of the lids under different environmental conditions and with different contents. We conclude that these jars form a safe, cheap, and laborsaving alternative for storage of alcohol collections as long as certain precautions are taken: 1) Check for corrosion by atmospheric humidity in the storage space. 2) To be on the safe side, use this kind of jar only for ethanol-preserved specimens. 3) Do not recycle a lid unless you are absolutely sure it is not damaged. 4) The lids are more resistant to higher concentration of ethanol than to lower ones. 5) Be sure that the lid is closed tightly.

INTRODUCTION

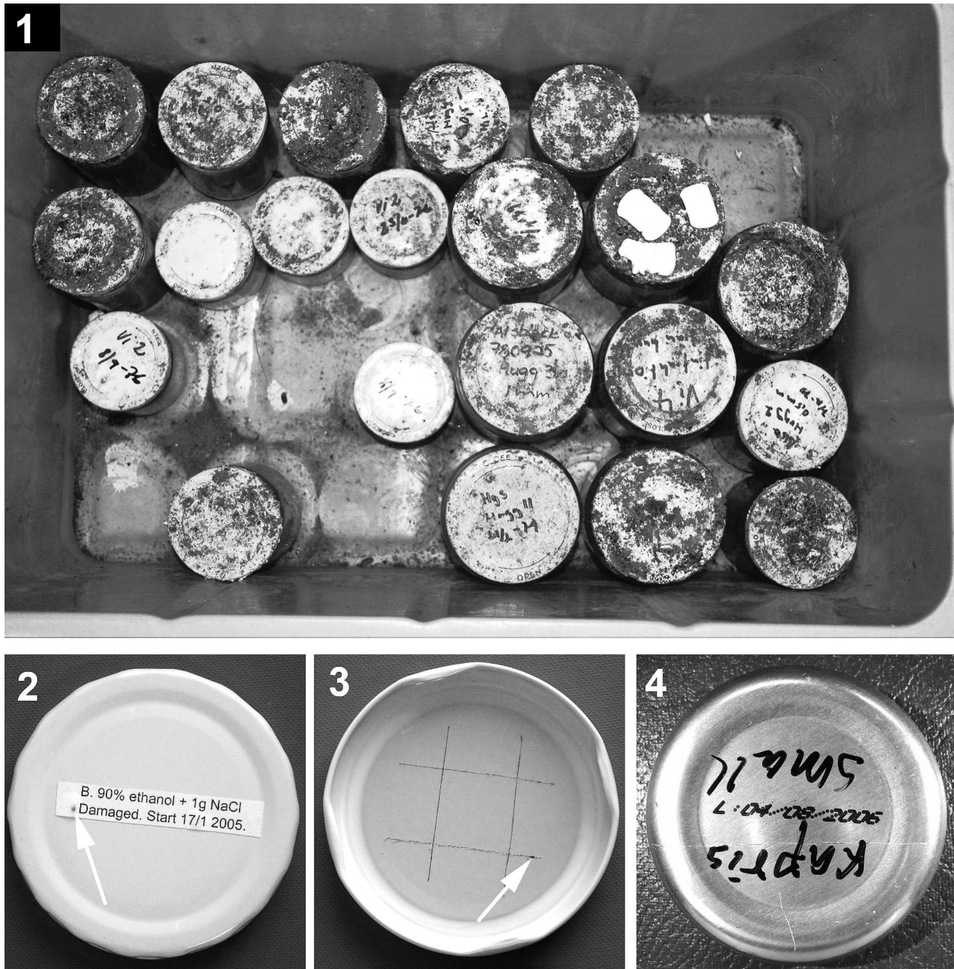
The Swedish Museum of Natural History (SMNH) has a large collection of animals preserved in ethanol, approximately 125,000 vessels with 375,000 lots, and some specimens, such as those illustrated by Seba (1734–1765) or used by Linnaeus, date back to the early 18th century. This historical significance adds a certain degree of responsibility for the storage and maintenance of our collections; at the same time the volume requires us to be efficient in our work.

Jars for storing alcohol preserved museum specimens seem to have been a problem as long as natural history museums have used ethanol for storage of specimens (Arndt 1937; Levi 1966; Clark 1993). During the last century, a number of closures were introduced: clamp lids, Bakelite lids (which quickly turned out to be unreliable), glass jars with polyvinyl-lined plastic lids, and various types of bail-top canning jars. The development and production of the Swiss Qualitight jars recently (Oberer 2001) seems to have stopped when the company went bankrupt in 2005, probably as a result of the high cost of the jars.

Clark (1993) reviewed the different types of glass jars that have been used at the Natural History Museum in London, and concluded that only glass jars with a ground-glass stoppers were, despite the high cost, suitable for long-term storage in museum collections. He considered that even if some other types could be used for a shorter time, most lids deteriorate and start leaking within 5–15 years. Clark also stated that metal lids easily get deformed or their inner liners become damaged, resulting in increased evaporation.

We largely share Clark's experiences, and agree that all Bakelite screw-on lids should be avoided, because often they crack and fit poorly, even when equipped with a liner, a conclusion also reached by Levi (1966). We share the experience that jars with ground-glass stoppers are very good, but the cost and risk of confusion of stoppers (which often are individually fitted) have prevented us from purchasing these for the SMNH. It also is common that the ground-glass lids get stuck, and require time-consuming work to be removed. Therefore, we are slowly recycling the best ones for historical material, but are replacing most with bail-top or metal lid jars.

We also have considered plastic-lid jars, such as the Fisherbrand™ series. The advantage of plastic lids is that they do not corrode. However, the cost is discouraging; it is several times that of bail-top jars, and the plastic lids are more difficult than metal lids



Figures 1–4. (1) Container with reference material from an environmental survey, transferred from the Swedish Environmental Protection Agency for storage at the SMNH. The contents of the jars are sometimes damaged by rust precipitations and occasionally dry. (2–3) External and internal views of a test lid intentionally damaged by four scratches on the inside. Note the small rust spot which has penetrated the label at the tip of the arrow. (4) Example of a metal-colored lid from the evaporation test in Table 2.

to close to a tight fit. Furthermore we do not know their longevity; we do have experience with a brand of plastic lids that started cracking more frequently after 20–30 years.

Concerned by having seen stacks of rusty and dry jars from old field work (Fig. 1), many collection managers categorically prohibit the use of jars with metal lids for storage of collections. Our experiences are more diverse. Most types of metal lids can be used for storage of alcohol-preserved material, but certain precautions should be taken to avoid corrosion or leaking. To avoid future failures, we have studied the behavior of the closures under normal storage conditions. We have also tried to accelerate their aging by storage at 50°C, a procedure often used in comparable studies by the governmental Technical Research Institute of Sweden to test material aging. Because 95% ethanol is increasingly used for at least short-term storage of specimens for DNA work (Carter 2002), we have tested that strength as well.

At the SMNH we have been using 390-ml jars with screw-on lids for some 25 years with no negative experiences. In Sweden they are sold by Nordic Pack (Box 4112, Klockarvägen 94, SE-151 04 Södertälje, Sweden, http://www.nordicpack.com/) as clear glass jar, Model 4390. The jars are marked with the letters "PLM" at the base and seem to be available in large areas of Europe from various glass distributors.

The lids come in two types, white and metallic golden (Figs. 2–4), and are 70 mm in diameter. The model designations are 70-2ME and 70-ME, respectively, and they are produced by Metropak AS (Rundageren 4, Postbox 180, DK-2640 Hedehusene, Denmark, http://www.metropak.dk/). A company representative states that the difference between the metallic golden and the white lid is that the latter has a double coat of paint to make it more resistant (Annelise Boyse, quality supervisor at Metropak, pers. comm. 5 October 2005), but no detailed description of paint and steel quality was obtained.

About 10,000 of these jars are now in use at SMNH, Department of Vertebrates and Department of Invertebrates. We have not been systematically recording damaged lids, but anecdotal memories indicate a few cases of such lids, which probably are only from formalin-preserved specimens.

Testing Methods

Our intentions in this study were 1) to quantify evaporation; and 2) to test the resistance of lids against corrosion, in order to get a factual basis for a decision about their continued use.

To quantify evaporation, we measured the loss of weight of jars filled with ethanol of different strengths by regularly weighing them for 3 years, with an accuracy of ± 10 mg. The jars were stored in our underground alcohol collection storage space at a temperature of 16–18°C and a relative humidity of 20–70%.

This test series was compared with two test series of French Le Parfait® Terrines, cylindrical bail-type jars with a volume of 1,000 ml used at SMNH for storing large specimens, and vials with minute specimens. These jars were equipped with two kinds of gaskets, the Le Parfait original, orange rubber gasket and a gasket from Luminarc®, another producer of bail-type jars (see www.leparfait.fr and www.luminarc.fr). The Luminarc gasket was called "Hifi" but unfortunately was replaced by another model, called "SiFacil" a few years ago. The Hifi gasket is by far the best standard gasket from a durability point of view and among the best for low evaporation. Although made for jars with a diameter of 70 mm (also the Hifi inner diameter), the Hifi gaskets can also be used for the wider, 85-mm Le Parfait Terrines. The new Luminarc SiFacil gaskets seem to fit only the corresponding Luminarc jars, because they contain an element of nonstretching plastic (the jars they are intended for are very good, although sometimes difficult to buy).

To assess internal corrosion, we prepared a series of tests of the inside of the lids of 60 jars. We tested alcohol concentrations of 40%, 50%, 60%, 70%, 80%, 90%, and 95% and 4% formalin (= formaldehyde solution). At each strength we used pristine lids, lids with scratches (in a "#" pattern, Fig. 3), jars with damaged lids, and 1 g sodium chloride (NaCl) to simulate dirty marine specimens. Two jars were used for each combination of concentration and physical condition. All dilutions were made with distilled water.

To test external corrosion we scratched the paint off several of the metal seating tabs that interlock with the ridges (threads) of the jar to tighten the lid (Fig. 10). This was supposed to simulate wear of the lids by repeated opening and closing. The test was

Table 1. Evaporation from fourteen 390-ml jars with ethanol 95%–40% at 16–18°C. Average evaporation was 0.16 g per jar and year. EtOH = ethanol.

		Date and Weight (g)									
Jar	% EtOH	18 January 2005	26 May 05	18 June 05	26 January 6	28 February 05	3 January 07	25 April 07	23 October 07	14 January 08	Total evaporation (g)
A	95	477.5	477.4	477.29	477.12	477.1	476.81	476.62	476.54	476.63	0.87
B	95	480.81	480.72	480.63	480.54	480.53	480.33	480.18	480.15	480.29	0.52
A	90	496.25	496.19	496.07	495.94	495.95	495.66	495.48	495.44	495.65	0.65
B	90	497.7	497.64	497.53	497.42	497.4	497.17	496.98	496.96	497.1	0.60
A	80	510.39	510.12	510.02	509.94	509.94	509.73	509.57	509.56	509.69	0.70
B	80	495.6	495.6	495.5	495.44	495.43	495.27	495.11	495.11	495.25	0.35
A	70	509.44	509.43	509.33	509.26	509.24	509.02	508.85	508.83	508.95	0.49
B	70	510.25	510.23	510.13	510.05	510.02	509.8	509.61	509.59	509.7	0.26
A	60	503.36	503.36	503.26	503.2	503.17	503.00	502.84	502.83	502.97	0.35
B	60	509.24	509.21	509.1	509.06	509.03	508.86	508.71	508.69	508.83	0.41
A	50	518.09	518.11	518.00	517.99	517.96	517.84	517.69	517.71	517.85	0.26
B	50	513.85	513.52	513.33	513.29	513.22	513.09	512.92	512.94	513.08	0.77
A	40	525.81	525.83	525.73	525.71	525.7	525.56	525.4	525.42	525.57	0.24
B	40	526.76	526.78	526.68	526.67	526.63	526.5	526.34	526.37	526.52	0.24

made with two jars each, filled with ethanol concentrations of 70%, 80%, 85%, 90%, and 95%.

To provide a baseline performance standard for metal lids in general and to determine the variation in evaporation among different brands, 12 jars of 10 different types were tested. The diameter of these lids varied between 42 and 70 mm. For this test we used 95% and 80% ethanol, and 33 months at a temperature of 50°C and 1 year at 16–18°C in the alcohol storage area.

For the formalin tests, the commercial solution of formaldehyde, ≈36% by weight, was diluted 1:9 to obtain a strength of ≈4% formaldehyde. In English and American literature this is often called 10% formalin because it is diluted to one-tenth of the original concentration.

Results

The evaporation from fourteen 390-ml jars was recorded by regularly weighing the jars that were stored in our alcohol collection (20–70% relative humidity and a temperature of 16–18°C) for 3 years. There was no significant difference in evaporation between 40% ethanol and 95% ethanol and the average evaporation (Table 1) was 0.16 g per jar per year and varied between 0.08 and 0.29 g per jar per year. This can be compared to 0.2–0.6 g, an average of 0.4 g per jar per year for bail-top Le Parfait jars, which is higher than that for the metal lids, but the diameter of the opening is larger.

To assess the variation between different types of commonly available jars with metal lids, we tested 12 jars with 80% and 95% ethanol in a well-ventilated incubator at a temperature of 50°C. After almost 3 the jars were moved to the alcohol magazine (relative humidity 20–70% and a temperature of 16–18°C) for a year to obtain a more practically applicable measure of the evaporation. The results are given in Table 2. The evaporation at 50°C was more than 10 times higher, compared with 16–18°C; i.e., 4.1 g per year, compared to 0.37 g in the alcohol storage. One jar, "Hallon," evidently had a problem (perhaps the lid had not been tightly closed), but if that one is omitted from the

calculation, the average evaporation at 17°C was 0.20 g per year, which is close to our results for the 390-ml jars and indicates that the metal lids have a fairly even quality, independent of brand or producer.

The corrosion test of the 390-ml jars used in our collections is presented in Table 3. Under the conditions in our alcohol collection, relative humidity 20–70% and a temperature of 16–18°C, we can conclude that:

1. No damage of the paint or metal was caused by atmospheric humidity.
2. No damage or rust occurred on the seating tabs where the paint had been scratched off. This agrees with our casual observations in the collection.
3. All jars with pristine lids had survived 3 years with no visible corrosion; this includes those used for 4% formaldehyde and 40–95% ethanol with NaCl added to simulate poorly rinsed marine animals.
4. The lids that had been intentionally damaged (Fig. 3) often were severely damaged. Important factors here were:
    a. Formalin is much more likely to corrode the lids than ethanol (Figs. 5, 8–9). In the test series with damaged lids, the rust spread laterally from the scratches and rust started to appear externally after only a month.
    b. Addition of NaCl to the ethanol sped up the corrosion process, and holes in the lids appeared after 4 months in 40% ethanol (Fig. 6).
    c. A high concentration of ethanol slows down the corrosion, but 95% ethanol might corrode the lids when they are damaged. However, after 3 years there were only traces of rust in the scratches.

One interesting observation is that the pattern of corrosion differs between formalin and ethanol. Ethanol starts the penetration of the metal on a very small surface and the penetration appears externally as a very small blister, 0.5–1.5 mm diameter (Fig. 2), whereas formalin penetrates over a larger area and in a more diffuse pattern.

Two jars with tap water were stored at 50°C for the same time as the main test. Their lids looked undamaged at a superficial inspection, but a closer examination showed that both lids were full of low blisters on the inside. Evidently the water had penetrated the plastic layer and had begun to damage the interior metal surface (Fig. 7).

Discussion

The results clearly show some important limitations of commercial jars with metal lids:

1. They are sensitive to any damage to the protective coating. This coating is thicker in a circular area where the lid is in contact with the glass. However, when used for field work, sediment particles easily are trapped between the lid and the glass, especially when shaking the jar to distribute the fixative or preservative. The next time the lid is opened and closed, the lid gets scratched on the interior surfaces (Figs. 8–9).
2. Formalin is more harmful to the lids than ethanol and the presence of electrolytes such as NaCl in the experiments or seawater in marine samples will increase the risk of damage. It is, therefore, important to rinse the specimens with 5–10% ethanol to wash out excess salt and formaldehyde prior to storage in ethanol. Use of fresh water can osmotically damage specimens, and strong ethanol does not dissolve salt as readily.
3. There is a risk of not closing the lid tightly enough, which requires some physical strength. Opening a lid after a few years requires more power, but can be facilitated by loosening it with a screw driver which also reduces the risk of accidental spilling of

Table 2. Mixed jars with metal twist-on lids, selected for size and form useful for invertebrate collections. Jars bought with content in supermarkets. Total evaporation 35.5 g per year or 4.1 g per jar per year at 50°C, 4.1 g per year or 0.38 g per jar per year at 17°C. Jumbo olives, lid not closed 13 September 07 and evaporation at 50° not included.

Name of jar	Date and Weight (g)									Loss in 17°C, 12 mo	Lid size and condition
	5 April 2004	14 April 05	26 August 06	15 April 07	30 June 07	13 September 07	3 January 07	Loss in 50°C, 33 mo	14 January 08		
Kapris small 95%	—	168.95	162.25	147.82	145.01	127.7	127.39	41.46	(Empty) 127.17	—	42.0 mm, few spots of rust
Kapris 130 mm 95%	—	259.96	258.6	255.21	255.02	249.36	249.14	10.82	248.94	0.20	56.0 mm, no damage
Olives 170 mm 95%	—	222.34	220.96	218.95	218.83	215.31	215.24	7.10	215.22	0.02	66.4 mm, no damage
Pesto 95%	—	126.76	124.53	122.38	122.07	112.08	111.88	14.88	111.86	0.02	49.5 mm, no damage
Woksås 95%	—	270.47	267.48	266.93	266.79	266.41	266.1	4.37	266.11	0.00	66.3 mm, some rust at edge
Jumbo olives 95%	—	391.67	387.12	372.06	371.36	303.83	303.27	—	302.37	0.10	66.5, small rust damage at edge
Sweet&sour 95%	290.53	290.43	288.59	283.56	282.68	373.5	273.22	17.31	272.79	0.44	55.8 mm, undamaged
Hallon 95%	343.64	332.62	331.87	328.97	328.71	322.22	321.32	22.32	319.12	2.20	69.0 mm, virtually undamaged
Lucullus 80%	100.53	100.29	99.55	97.7	97.11	94.07	93.92	6.61	93.75	0.17	41.8 mm, possibly 1 blister
Lucullus 95%	91.83	91.59	90.1	86.78	85.75	80.38	80.11	11.72	79.79	0.32	41.8 mm, undamaged
Paradiso oil 80% 170 mm	—	—	—	—	—	—	399.54	—	399.17	0.37	66.4 mm, a few very small spots
Figaro oil 80%	—	—	—	—	—	—	208.55	—	208.26	0.30	44.6 mm, a little rust on thick part
Total evaporation (g)	—	—	—	—	—	—	—	**95.13**		**4.14**	
Temperature (°C)	50	50	50	50	50	50	50	Transfer to 17	17		

Table 3. Test of corrosion started 17 January 2005. Jars with ethanol: + = small, single speck of rust; ++ = speck of rust (in scratches); 1,2 = rust in large part of scratch; 3–6 = lid penetrated by rust. Jars with formalin: 7 = scratches covered by laterally spreading rust; 8–9 = scratches start to disappear; 10 = scratches not discernible.

				Date checked									
Content	Jar	Added NaCl	Lid, start condition	24 February 2005	18 March 2005	18 April 2005	25 May 2005	17 June 2005	6 September 2005	3 January 2007	25 April 2007	23 October 2007	18 January 2008
% EtOH													
40	A	—	Scratched	++	++	++	1	1	1	1	1	1	2
40	B	—	Scratched	+	++	++	1	1	1	1	1	1	2
40	A	1g	Scratched	1	1	2	2	3	4	6	6	6	6
40	B	1g	Scratched	1	1	1	2	2	3	3	4	4	5
40	A	—	Whole	0	0	0	0	0	0	0	0	0	0
40	B	—	Whole	0	0	0	0	0	0	0	0	0	0
40	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
40	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
50	A	—	Scratched	0	+	+	+	+	1	1	1	1	1
50	B	—	Scratched	0	0	0	0	0	0	2	2	2	2
50	A	1g	Scratched	1	1	1	1	1	2	3	4	4	5
50	B	1g	Scratched	1	1	1	1	1	2	4	5	5	5
50	A	—	Whole	0	0	0	0	0	0	0	0	0	0
50	B	—	Whole	0	0	0	0	0	0	0	0	0	0
50	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
50	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
60	A	—	Scratched	+	+	+	+	+	+	+	+	+	+
60	B	—	Scratched	++	++	++	++	++	1	1	1	1	1
60	A	1g	Scratched	+	1	1	1	1	1	3	4	4	5
60	B	1g	Scratched	++	1	1	1	1	1	4	4	4	5
60	A	—	Whole	0	0	0	0	0	0	0	0	0	0
60	B	—	Whole	0	0	0	0	0	0	0	0	0	0
60	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
60	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
70	A	—	Scratched	++	1	1	1	1	1	2	2	2	2
70	B	—	Scratched	++	++	++	++	++	1	1	1	1	2
70	A	1g	Scratched	++	1	1	1	1	1	4	4	4	5
70	B	1g	Scratched	1	1	1	1	1	1	3	3	3	3
70	A	—	Whole	0	0	0	0	0	0	0	0	0	0
70	B	—	Whole	0	0	0	0	0	0	0	0	0	0
70	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
70	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
80	A	—	Scratched	0	0	0	0	0	0	0	0	0	0
80	B	—	Scratched	0	0	0	0	0	0	++	++	++	++
80	A	1g	Scratched	1	1	1	1	1	1	3	3	3	4
80	B	1g	Scratched	1	1	1	1	1	1	3	3	3	4
80	A	—	Whole	0	0	0	0	0	0	0	0	0	0
80	B	—	Whole	0	0	0	0	0	0	0	0	0	0
80	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
80	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
90	A	—	Scratched	0	0	0	0	0	0	2	2	2	2
90	B	—	Scratched	0	0	0	0	0	0	2	2	2	2
90	A	1g	Scratched	+	++	1	1	1	1	2	2	2	2
90	B	1g	Scratched	++	1	1	1	1	1	2	2	2	2
90	A	—	Whole	0	0	0	0	0	0	0	0	0	0
90	B	—	Whole	0	0	0	0	0	0	0	0	0	0
90	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
90	B	1g	Whole	0	0	0	0	0	0	0	0	0	0

Table 3. Continued.

				Date checked									
Content	Jar	Added NaCl	Lid, start condition	24 February 2005	18 March 2005	18 April 2005	25 May 2005	17 June 2005	6 September 2005	3 January 2007	25 April 2007	23 October 2007	18 January 2008
95	A	—	Scratched	0	0	0	0	0	0	++	++	++	++
95	B	—	Scratched	0	0	0	0	0	0	0	0	0	0
95	A	1g	scratched	+	+	+	+	+	+	++	++	++	++
95	B	1g	Scratched	0	0	0	0	0	0	0	0	0	+
95	A	—	Whole	0	0	0	0	0	0	0	0	0	0
95	B	—	Whole	0	0	0	0	0	0	0	0	0	0
95	A	1g	Whole	0	0	0	0	0	0	0	0	0	0
95	B	1g	Whole	0	0	0	0	0	0	0	0	0	0
% Formalin													
4	A	—	Scratched	7	8	8	8	8	9	10	10	10	6
4	B	—	Scratched	7	8	8	8	8	9	10	10	10	6
4	A	—	Whole	0	0	0	0	0	0	0	0	0	0
4	B	—	Whole	0	0	0	0	0	0	0	0	0	0

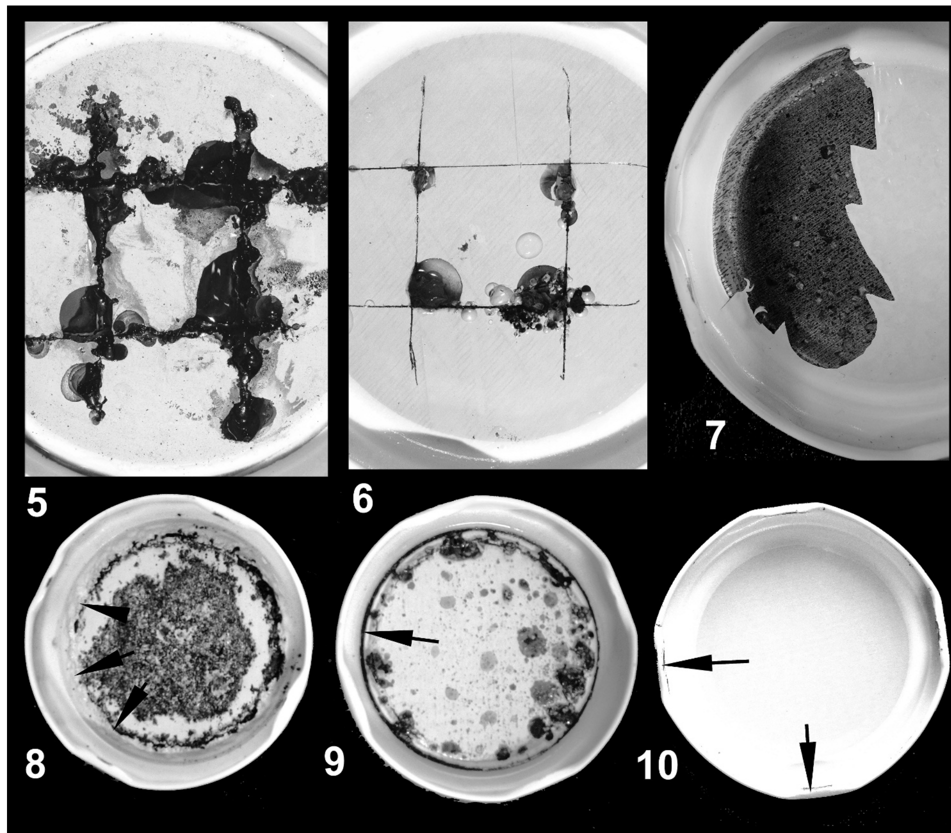
ethanol. For a 70-mm lid, a screw driver with a tip of about 4 × 1 mm can be inserted between the lid and the jar and twisted slightly in a couple of places to facilitate unscrewing the lid. Lids that have been removed this way should never be reused.

The problem with the damage to the lids, however, is counteracted by one of the advantages, the low cost. We pay roughly €2.50 for a 1,000-ml Le Parfait jar plus €0.15–0.20 each for gaskets from an alternate source; the 390-ml jars cost €0.15 and a lid costs €0.05. Therefore, lids can and should be discarded at the slightest suspicion of a problem. The lids are sold separately, and for the invertebrate collections we have a spare supply of three times the number of jars in use. The jar model and the lid have remained unchanged for at least 30 years.

Even when a lid rusts through, there is no immediate catastrophe. A minor rust hole is not totally open and might not expose the jar contents and, according to preliminary observations, it will take several months or years for the alcohol to evaporate.

One undisputable advantage of metal lids is the high reliability (as long as the aforementioned weaknesses are considered). Additionally, there is a low level of evaporation, which is on an average 0.16 g per jar per year (range = 0.08–0.29 g) and better than the Le Parfait jars at approximately 0.32 g per jar per year (range 0.21–0.61 g; Tables 4–5). A part of the explanation for low evaporation, compared to plastic and Bakelite lids, might be that the friction between the lid and the jar is lower than that between plastic or Bakelite and glass and makes it possible to close the metal lid better.

Jars with polyvinyl-lined plastic lids have the obvious advantage of eliminating corrosion, whereas metal lids are sensitive to high ambient humidity. In our underground storage space, the relative humidity varies between 20% and 70% during the year, due largely to seasonal variation, but we have seen no sign of external corrosion of the lids. For the evaporation from plastic lid jars we do not have any long-term measurements, only a short series (in progress), which indicates higher evaporation than in metal-lid jars. The deciding factor was the price, which is five times higher for plastic lids.



Figures 5–10. (5) Inside of a damaged lid from a jar containing 4% formaldehyde solution. (6) Inside of a damaged lid from a jar containing 40% ethanol with 1 g of NaCl added. Figures 5–6 were photographed at the same occasion, one year after start. (7) Inside of a jar containing tap water after 3 years storage at 50°C. The internal side of the lid is covered with blisters underlain with rust, and the internal plastic coating is partly pulled off where it was peeling. (8–9) Two undamaged jars used for storage of 4% formaldehyde solution for 3 years at 50°C. Notice the circular damage pattern from contact of the lid with the jar (arrow). In (8), the central part of the lid is held in place only at the three arrows. (10) Test lid with damaged seating tabs (arrows) after 3 years with 40% ethanol.

Conclusions

Metal-lid jars only should be used for short-term storage of formalin-preserved material. For long-term storage they are suitable only for alcohol-preserved specimens, and with the following precautions:

1. Be absolutely certain that the lid is intended for and the right size for the jar.
2. The lid should have internal paint or a plastic coating thick and durable enough to resist some wear. Never use a lid that has a scratch. The lids should also have an extra layer of sealant inside to function as a gasket. Never use metal-colored lids.
3. Do not tighten the lid excessively. The seating tabs of the lids can be bent so they no longer hook on to the thread of the jar and, therefore, do not seal well. A certain degree of deformation of the lid actually promotes a tight seal.
4. Lids are inexpensive; discard them at the slightest suspicion of something wrong.

Table 4. Six Le Parfait Terrines, 1,000-ml jars with orange Le Parfait gaskets and Luminarc Hifi gaskets. Long-term comparison of evaporation with 500 ml 80% ethanol at room temperature over 8.5 years. Total evaporation = 18.9 g 80% ethanol. Condition of gasket: * = outside of gasket slightly harder, inside not yet soft and sticky; ** = no change. EtOH = ethanol.

	Date and Weight (g)					
Le Parfait Jars	5 July 1999	26 March 2002	6 April 2004	19 January 2008	Evaporation (g) and gasket condition	Loss per year (g)
Original orange gaskets						
1	1,215.5	1,214.9	1,213.5	1,211.4	4.1 *	0.48
2	1,210.4	1,210.1	1,209.0	1,207.0	3.4 *	0.40
3	1,210.4	1,210.0	1,209.4	1,208.1	2.3 *	0.27
Hifi gaskets						
1	1,206.7	1,205.4	1,203.9	1,201.6	5.1 **	0.60
2	1,227.8	1,227.5	1,226.7	1,225.5	2.2 **	0.26
3	1,224.4	1,224.0	1,223.6	1,222.6	1.8 **	0.21

Table 5. Short-term test of evaporation at room temperature from a series of 1,000-ml Luminarc French canning jars with silicone rubber Hifi gaskets. EtOH = ethanol.

		Date and Weight (g)								
Jar	% EtOH	26 May 2005	18 June 2005	26 January 2006	28 February 2006	3 January 2007	25 April 2007	14 January 2008	Evaporation (g)	Loss per year (g)
A	80	1,541.7	1,541.4	1,541.4	1,541.4	1,540.9	1,540.7	1,540.6	1.2	0.46
B	80	1,549.6	1,549.2	1,549.3	1,549.3	1,548.7	1,548.6	1,548.5	1.1	0.42
C	80	1,540.6	1,540.3	1,540.2	1,540.2	1,539.6	1,539.4	1,539.3	1.6	0.61
D	80	1,545.1	1,544.7	1,544.8	1,544.8	1,544.2	1,544.1	1,544.0	1.1	0.42
E	80	1,543.2	1,542.8	1,542.9	1,542.8	1,542.3	1,542.2	1,542.1	1.2	0.46

5. Test a new brand of jar for a few years before taking it into general use.
6. Purchase at least 100% more lids than jars. Lids disappear or are damaged; the jars remain.
7. Under careful supervision, test the lids under both real conditions and with simulated damage.

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DEVELOPMENT OF BEST PRACTICES FOR INTEGRATED PEST MANAGEMENT AND A BEST PRACTICES MODEL FOR THE WIDER MUSEUM SECTOR

Lisa Kronthal Elkin,[1] Christopher A. Norris,[2] and Derya Golpinar[3]

[1]*Natural Sciences Conservation Laboratory, American Museum of Natural History, Central Park West at 79th Street, New York, NY 10024*
[2]*Division of Vertebrate Paleontology, Yale Peabody Museum of Natural History, 170 Whitney Avenue, P.O. Box 208118, New Haven, CT 06520*
[3]*Lower East Side Tenement Museum, 91 Orchard Street, New York, NY 10002*

Abstract.—The Integrated Pest Management Working Group (IPM-WG) was formed in 2002 as an ad hoc group of museum professionals dedicated to the development of pest management resources for the general museum community. Through its Standards and Best Practices (S&BP) Subgroup, the IPM-WG has been evaluating pest management policies and procedures from a variety of cultural institutions with a view to developing policy and procedural templates that can be employed in the development of integrated pest management programs. In pursuing this goal, the S&BP Subgroup first considered the mechanisms by which standards and best practices in pest management could be developed and used. This information was distilled into several tools and templates for distribution to the wider museum community. We believe the process adopted by the IPM-WG could be applied to other areas of museum practice as a model for the development of best practices.

Introduction

Best practices have been variously defined as techniques or methodologies that, through experience and research, have proven to reliably lead to a desired result (SearchSoftwareQuality 2007); procedures that are generally agreed upon but not legislated (Cato et al. 2003); or commendable actions and philosophies that successfully solve problems, can be replicated, and demonstrate an awareness of professional standards (Merritt 2008). Best practices commonly are linked with standards. Unlike standards, however, best practices cannot be enforced or set easily as a target for attainment. Merritt (2008) characterizes best practices as being ''extra credit,'' when compared to the fundamentals represented by standards; museums should be applauded for achieving them, but not faulted if they can't. Some best practices might not be suitable to the particular circumstances of a museum, whereas others might be beyond the resources available (Merritt 2008).

Codification and development of best practices generally are seen as desirable goals for the museum sector (e.g., Macklin 2010). However, there are a number of significant challenges to achieving these goals. To be maximally effective, best practices must be drawn from the widest possible sample of community procedures. The assessment of these procedures needs to be undertaken critically by an adequate cross-section of the professional community. The best practices developed from this process should be made available to the community through publication, either via print or the World Wide Web. Finally, because best practices are subject to continual refinement and evolution, there must be mechanisms in place for community feedback and regular review.

Traditionally, the role of developing and promulgating best practices has been taken on by professional societies (Macklin 2010) on the basis that these bodies are best placed to access the collective knowledge of their communities and to draw on this knowledge for critical assessment. Best practices that are developed in this way come with a stamp of approval from the society taking the lead, giving them added weight. Under certain

circumstances, however, the focus of individual societies might be too narrow to adequately sample the widest possible range of expertise. One such area is that of integrated pest management (IPM).

IPM, defined as the selection, integration, and implementation of pest management methods based on predicted consequences (Cato et al. 2003) is a necessity for a wide range of cultural heritage institutions, including museums, galleries, libraries, archives, and university and private collections. It extends across a range of operations for these institutions, including not just collections care, but also facilities management, custodial services, retail and catering, and finance. IPM draws on the expertise of a number of different professional groups, including curators, collection managers, conservators, registrars, facilities managers, maintenance and custodial staff, and administrators. The ultimate objectives of these groups can differ substantially and coordination of activities is critical for successful pest management.

It was in recognition of these challenges that the IPM Working Group (IPM-WG) was founded in 2002. The IPM-WG is an ad hoc grouping of professionals from both the cultural heritage community as well as the private sector who have an interest in pest management issues. The IPM-WG focuses its efforts on a number of thematic areas, each covered by a subgroup of IPM-WG members. One of these subgroups was focused on the dissemination of best practices in IPM. The creation and activities of this subgroup represents a potential model for the development of best practices in the wider museum sector.

Background to the Group

The IPM-WG arose from an informal meeting held at the American Museum of Natural History (AMNH) in the Fall of 2000 among staff from AMNH, the Museum of Texas Tech University, the National Museum of the American Indian, and the Canadian Conservation Institute who were working on developing databases for use in recording and mapping pest outbreaks in museum collections. This information-sharing exercise ultimately led to a more formalized meeting looking at widespread community needs in integrated pest management, which was held at AMNH in 2002. This was the first meeting of the IPM-WG, which has met annually at AMNH since then. Currently the group has over 70 members representing more than 30 different cultural institutions from both Europe and North America. The membership covers natural sciences, arts, humanities, libraries and archives, and is drawn from a mix of state and federal agencies, universities, stand-alone institutions, and commercial vendors. Individual members come from a variety of different professions, including conservators, collection managers, curators, entomologists, facilities managers, librarians, and archivists.

Membership of IPM-WG is open; anyone with an interest in pest management in cultural institutions is free to attend the group's annual meeting, provided they are prepared to work in support of the group's objectives during and after the meeting. Attendees are selected on a first-come, first-served basis; there is a limit on attendee numbers imposed by the venue, and in most years requests to attend have exceeded the number of places available.

In addition to the annual meeting, the IPM-WG offers a number of resources to the heritage preservation community. Principal among these is a Web site, MuseumPests (www.museumpests.net), which provides a wide range of downloadable resources for pest management. The IPM-WG also runs an email listserve focusing on pest management issues that currently has just over 600 members, and maintains a login WIKI site to facilitate the work of Group members.

At an early stage in the development of the IPM-WG, the membership debated the issue of whether to seek formal affiliation with a professional society. Ultimately, it was decided that there was no way to do this without potentially limiting either the scope of the resources produced, or the range of membership present in the group. Up to 2009, the IPM-WG remained an ad hoc grouping, hosted by AMNH and receiving a small amount of commercial sponsorship, with members and their home institutions covering the costs of meeting attendance. In 2009 support for upgrades to the MuseumPests Web site was sought and obtained from a number of professional organizations and institutions (see acknowledgments for full listing).

Early on, the Group decided to focus its efforts in five thematic areas, each of which would have its own subgroup. These were:

1. **Data collection**, which covered monitoring and trapping methodologies, as well as record keeping and reporting, data modeling, and database development;
2. **Identification Aids**, which concentrated on imaging and developing resources such as fact sheets, image libraries, and identification resources for common museum pests;
3. **Treatments**, which compiled bibliographies, case studies, and methodologies for both chemical and non-chemical treatment options, including heat, freezing, and anoxia;
4. **Web Resources**, which provided a compilation of useful resources related to IPM and also oversaw the development of the MuseumPests Web site; and
5. **Standards and Best Practices**, which focused on three areas—IPM policy development, procedures for putting policies into practice, and tools (such as education packages, risk and cost analyses, and scientific studies) for supporting policies and procedures.

Each group amassed bibliographies, references, courses, and workshops related to its area. This paper focuses on the work of the last of these subgroups, Standards and Best Practices (S&BP).

Operation of the S&BP Subgroup

Early on, the S&BP Subgroup concluded that it was not sufficient to simply develop the best practice elements for an IPM plan. In order to fully realize the potential of these elements as a community resource, collection professionals would also need access to tools that would enable best practices to be implemented. To do this, three questions needed to be answered: who is the potential audience, what resources are already out there, and what can we add?

The Audience: The IPM Grid

The importance of identifying the potential audiences for future policy or procedure documents that might be produced and distributed by IPM-WG cannot be overstated. Seven institutional "supergroups" were defined. All of these would need to be involved in the implementation of an institution-wide pest management plan and for the plan to be a success they would all need to be influenced to some extent. These groups are listed in Appendix 1.

The ability of any one staff member, or group of staff members, to directly influence all of these groups was likely to be limited. Success was dependent on: 1) knowing the role of each group in the context of IPM, 2) finding their motivation for participation, and 3) speaking to their particular need.

The first of the S&BP Subgroup's products, the IPM Grid (Appendix 1), arose directly from this conclusion. The grid was a matrix developed by the S&BP SUBgroup for the

delivery of information. On its vertical axis, the subgroup identified categories of people/audiences within an institution that would need to be involved and influenced in order for an IPM plan to be successful. The horizontal axis showed examples of the responsibilities of these groups, and outlined some of the arguments and incentives that might be used to secure their cooperation. The grid is not a best practices document per se, but it does lay out the framework of consultation that is necessary to implement an institution-wide IPM plan. In doing so, it laid the groundwork for the subgroup's development of best practices documents.

Vetted Documents—Policies, Procedures, and Tools

For each of the three areas within S&BP—policies, procedures and tools, the subgroup then concentrated on gathering available resources from the community; this was done through a combination of Web searches, gathering existing material from members' home institutions, and calls for submissions sent out via listserves. These documents were vetted by the subgroup on an annual basis, during the IPM-WG meetings, with the aim of creating a set of suitable examples for distribution via the MuseumPests Web site.

For the vetting process, the S&BP Subgroup pulled together a set of 46 institutional documents relating to IPM. These documents were filtered into three categories—policies, procedures, and tools—and evaluated for posting. The subgroup reviewed each document for content, based on initial discussions on what topics should be included in a policy. Wherever possible, examples were chosen that would have broad applicability, rather than those that were very institution or collection specific. By the end of this process, the S&BP SUBgroup had selected a group of documents that the subgroup believed would provide a good set of examples to underpin development of an IPM program. After obtaining permission from the institutions concerned, these were posted on MuseumPests.net along with abstracts written by subgroup members.

Templates—Policies and Procedures

The strongest elements of the selected documents were incorporated into a template policy (and ultimately procedure) document, also made available via the Web site. Based on the document review, the subgroup was able to identify a collection of elements or statements that collectively represented a set of minimum requirements for an IPM policy. These elements were:

- Introduction: A statement of what the document is—in this case, a pest management policy.
- Objective/Scope: What outcome(s) is the policy intended to lead to?
- Justification: Why is the policy needed?
- Applicability: Who will the policy cover? Who will have overall responsibility for its implementation? Who else has roles that are defined by the policy?
- Training: What are the arrangements for training staff to make them aware of the policy and their responsibilities under it?
- Support/Budget: A statement that implementation of the policy is supported at an institutional level and that an appropriate budget will be made available for implementation.
- Best practices: A statement explaining how the policy follows the appropriate best practices for the discipline(s) concerned.
- Monitoring: How will the effectiveness of the policy be measured?

- Remediation: In the event of a pest outbreak, what steps will be taken to contain and treat the outbreak?
- Documentation: A list of other institutional documents relevant to the policy.
- Review/Revision: What are the procedures and timescale for reviewing the effectiveness of the policy and making revisions if necessary?

Using this list of elements, the S&BP SUBgroup was able to generate a template for writing an IPM policy (Appendix 2). Each item on the list forms a section heading with accompanying guidance notes for writing the policy that were produced by the subgroup. The notes are based on the ideas about framing messages to meet the needs, concerns, and prejudices of the relevant institutional supergroups that are set out in the grid. After a few rounds of amendments, the policy was approved by the subgroup at its meeting in February 2008 and posted on MuseumPests.net.

The subgroup used the same process of document distillation to create a series of templates from the procedure documents that also were reviewed and vetted by the subgroup members. Many of the elements were shared with the policy template, the main difference being information specific to individual procedures. Individual subgroup members were tasked with drafting templates that were then discussed and edited by the wider group. As with the policy template, the S&BP SUBgroup used the grid to ensure that the documents spoke effectively to the relevant institutional players. The end result was an initial set of six procedural template documents:

- Preventing Access for Pests;
- Housekeeping;
- Control of Food and Live Plants;
- Control of Climate and Water Sources;
- Monitoring, Data, and Analysis; and
- Remediation.

These were approved by the subgroup and posted on MuseumPests.net.

Conclusions

Several lessons were learned about developing best practices from the IPM-WG, specifically with regard to Standards and Best Practices efforts. First, one of the strengths of IPM-WG is that it draws from a wide range of experiences and expertise. Many different professions, different disciplines, and different types of institutions are represented by the group's membership. This breadth of expertise is vital for the development of best practices and provides a potential step towards the community/audience buy-in required for the eventual development of professional standards in this area.

Secondly, this was a bottom-up or "grass roots" process, which was community-led and -supported. The IPM-WG is not affiliated with any particular institution or professional society. Although AMNH provides a small amount of logistical support, and the group has received some sponsorship from professional societies and from industry for upgrades to the Web site, the bulk of the costs of the group, which are associated with attending the meeting, are met by participants or their home institutions. This gives members of the group a genuine sense of ownership for the resources that they have produced.

Thirdly, this is an open process. A general invitation to the meeting is sent out via listserves. Anyone who wants to attend is free to do so. The work processes for the IPM-WG all are fully transparent. During the drafting process, documents are posted for comment and review on the group's WIKI site and the final documents are made available to the wider community via the MuseumPests Web site. Another important feature of the IPM-WG's work on best practices is that throughout the process the focus has been a practical one; to develop tools and resources that can be downloaded and used by any institution.

The work of the Standards and Best Practices Subgroup of the IPM-WG provides an example of how standards and best practices can be framed in a community-led process that goes across traditional institutional and disciplinary boundaries. The development of the grid as a first stage enabled the subgroup to frame documents that would speak directly to the major institutional players in any IPM plan. We believe this approach is widely applicable to standards and other policy development across the museum sector.

Acknowledgments

The authors would like to extend their thanks to the many members of the IPM Working Group who contributed to the assessment of documents and development of the templates, especially Rachael Perkins Arenstein, Neil Duncan, and Richard Monk. Support for the redesign of the MuseumPests.net Web site was provided by the Objects, Conservators in Private Practice, Wooden Artifacts, Book and Paper, and Paintings Specialty Groups of the American Institute of Conservation; the American Museum of Natural History; Artex Fine Art Services; the Denver Museum of Nature and Science; Insects Ltd. Inc.; the Museum of Fine Arts, Boston; the Society for the Preservation of Natural History Collections; Historic New England; and Zak Software Inc.

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Appendix 1.

MuseumPests.net

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Standards & Best Practices Grid

This document is a matrix mechanism to help people developing IPM plans target information to the "super-groups" involved in the funding and execution of these plans. On the vertical axis, the grid identifies the categories of people within an institution who have to be influenced in order for a plan to be successfully implemented. Along the horizontal axis are examples of the responsibilities of each of these groups, and the sort of arguments and incentives that can be deployed in each case to ensure their cooperation.

Super-Group	What should they do for Collections IPM?	What is their incentive?	What tools/arguments can we use to affect this incentive?
Administration	Identify IPM as an institutional priority, this should get up to the level of trustees and then to all staff. Ties to city government could be effective through local sanitation and health regulations. Allocate funding to get IPM on capital plans for infrastructure/repair, monitoring services, education. Reinforce primary IPM nature of custodial work. Recognize hidden IPM threat and subsequent costs in events & construction.	Allocation of funds to support an essential and effective IPM program with measurable results should convince them that this will save them money. Convince them that they're preserving collections value & saving staff time (triage). Improve staff health by reduction in pesticide use, allergen exposure, pest borne disease. Recognize public relations issues with infestation such as passing pests on to other institutions, health audits, and accreditation.	Examples of successful studies Cost Analysis of IPM activities. Risk Assessment. Quantify: IPM time allocation, hazards learned from trapping and inspections, history and extent of damage to collection by pests, remediation costs.
Building Management (custodial, HVAC, construction, grounds, shipping)	Custodial Services (this is the primary line of defense) – reporting pest sightings (with basic training), identify frequency of duties by zone, more involved in monitoring. Construction - pest management clause in contracts (seal off affected area to a standard, cleanliness/sanitation levels, etc.) Shipping – monitor/look for problems, reporting, know regulations. Grounds – knowledge about exterior plant design, choice of plants, reporting. HVAC – window policies, opening screens, filtration standards, maintenance. Exterior door seal choices, installation and maintenance, bird nettings etc.	Elevated status for custodial in eyes of administration. Construction leaves institutions vulnerable to pests (and other hazards). Administration will want Building Management to exercise due diligence in protecting the collection from harm. International traffic in pest organisms attached to goods is becoming more regulated as it is recognized as a serious economic hazard. Crating and shipping staff will be responsible for meeting regulations. Some pests are indicators of building mould problems. Mould remediation is a costly problem. Early detection of building mould is a potential cost savings. Prevention preserves human health.	Training packages showing what is needed from building management for effective IPM. This is what could happen if we don't get it. How IPM coordinates with mitigating other hazards. Develop templates of IPM related inclusions for contracts.
Security & Safety	In their role of visitor and staff control, security performs policy enforcement and reporting functions. IPM needs to be tied more intimately to security for the following benefits: keeping doors closed, noticing pest activities at night, restricting food use to allocated places. Safety: fumigation alarms tied to security system alarm panel, freezer alarms, monitor external	IPM offers a lowering of use of hazardous chemicals, restriction of pesticides and the reduced need for giving applicators access to collections areas. Security and IPM can assist each other by staying current on legislation and providing safer alternatives to the institution. Help with handling issues.	Develop training materials on basic IPM awareness, identification, and useful contribution through reporting pest sightings. Link to sources of fumigant transport, use and pesticide use regulations and develop template policies to ensure artifact safety.

Super-Group	What should they do for Collections IPM?	What is their incentive?	What tools/arguments can we use to affect this incentive?
	door closure (seal against miscreants and pests). Hazardous materials (pesticides, fumigants) often under security concern as 'Director of Safety'.		
External Vendors (food services, events, shops)	Follow wording of contracts (with vendors, catering, etc.) Report detection of pest problems with their inventory or locale. Follow IPM requests for events, facilitate custodial activities.	A good working relation between institution and the vendor will lead to future business. An informed vendor, sensitive to institutional concerns is more likely to be engaged. The institution should be equally gracious in any forthcoming information on revealed pest problems.	Develop contract templates to note proscribed behaviors, timely clean up, etc. Develop training materials for events that deal with proper quarantine inspection of incoming goods and post event handling of flowers, garbage, prop materials, etc. Integrate with custodial training.
Collections/Research (staff, researchers)	Include IPM in loan policy, visitor policy, treatments. Reporting function - external researchers, contractors, visitors – they're the ones using the collections. Separate offices from collections to zone areas of IPM control.	Do unto others... Collections staff should be the most sensitive to pest hazards, and are the most exposed to pest control methods, historical or presently used. Professional standards should ensure that modern IPM methods are incorporated into everyday practice.	Training staff & researchers in basic IPM principles and local practices, hazard awareness and handling precautions to protect them against pesticide residuals. Develop example IPM programs that can be adopted and adapted by line collection staff in various collection types.
Exhibitions & Education	Education –could be a partner in what we're doing in IPM education. Teaching collections need to follow same standards as main collections, but often with more diligence due to risk (hopefully well controlled) of exposure to untrained public and short term volunteers (e.g., arsenic treated specimens). Exhibitions contract/loan issues, case design integrating IPM principles (insect barrier, access for inspection, no hidden areas, etc.), maintenance, on site construction, IPM needs to be part of the design process and have a review function in a positive role from the outset, rather than a solely remedial role.	Ability to use collections safely in the course of their professional programs, extended ability to showcase 'backroom' collection care methods to the public. Impart a sense of ownership to visiting public in reducing the hazard from pests to augment the security role in enforcement of policies (example: food use).	Develop training package for education staff on IPM principles that could be applied to household use, and interpretation material on museum pests and how modern museums cope with them in a 'low impact' way. Suggest IPM program for interpretive support materials, such as dress up clothing (example: awareness of lice control methods) and collection elements.
Human Resources	Staff training could occur at orientation (volunteers, interns, students, staff).	Assisting professional staff development is part of their job. Knowledge of professional competencies is part of their managing staff hiring and promotion processes.	Develop explanations on IPM functions and suggest sample IPM related inclusions for job descriptions.

APPENDIX 2.

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POLICY DOCUMENT TEMPLATE

- *This document is a template to assist you in writing IPM policy documents for your collection and/or institution.*
- *If you already have a standard institutional policy format, then you need to make sure that your policy follows that formatting.*
- *Remember that if your institution already has a collection management policy or other relevant policy documents in place, you must be careful to ensure that your IPM policy does not conflict with these.*
- *Policy documents frequently require approval at the Board level and so should be as concise as possible – wherever possible, you should avoid talking about the details of how the policy will be implemented (these should be covered in a separate procedures document).*
- *The section headings in this template are primarily for your guidance in setting out the main topics that will need to be covered. You may choose to dispense with section headings altogether in your document, or to limit their number.*

Objective *[or]* Scope

This should be a brief statement setting out the scope of the document. For example, a food control policy document might begin with a statement that "This document is intended to set out the policy of [institution name] regarding the consumption of food and drink in public and non-public areas."

Introduction *[or]* Justification

This part of the document should briefly define what IPM is, and why pest management is important for your collection and/or institution.

Applicability

This section should set out whom the policy will apply to. The document should clearly identify a staff member who will have primary responsibility for ensuring compliance with the policy for the collection and/or institution. This person will delegate accordingly.

This section should also include a general statement of the roles and responsibilities of staff, volunteers, contractors (including caterers) in respect of the policy. More detailed information on individual responsibilities should be covered in procedure documentation.

Support [and/or] Budget

In this section, include a statement that the institution will provide appropriate support for the pest management activities of its staff and that adequate funding will be allocated. You may also specify the person or group responsible for ensuring that such funds are available.

Training

This section should emphasize the need for staff to go through regular and appropriate training; it may be as brief as a short statement to this effect. If a training program is to be set up as part of the policy, this should be noted. Details of training should be dealt with in procedure documentation.

'Best Practices'

This section should include a statement that appropriate control measures and programs will be put in place. If there are particular community-wide professional standards or best practices that apply to this policy document, they should be noted here as part of a statement that the institution will at all times follow them in the application of the policy.

Monitoring

This section should not contain the details of the monitoring program (e.g. trapping, observation, etc.), which should be covered in procedures documentation, but a statement of how the efficacy of the policy will be assessed.

Remedial Action

This will be a statement of the actions to be taken to remedy failures of the policy – e.g. dealing with a pest outbreak. If you want to enforce a policy that specifically excludes chemical treatments, this is where it should sit. As before, you should avoid going into specifics, which can be covered in procedural documentation.

Documentation

A list of the documentation that is linked to the policy may be included in this section of the policy along with 1) other relevant institutional policy documents relating to IPM, collection management, security, etc. 2) procedural documents that lay out the specifics of the policy's implementation and 3) other relevant documents external to the institution, e.g. relevant professional standards.

Revision/Review

It is important to revisit the policy at regular intervals to ensure that it is up to date and to incorporate amendments that have arisen from your experience of implementing the policy. This section of the policy should specify the intervals at which the policy will be reviewed, who will be responsible for initiating the review, and who will be involved.

CONDUCTING COLLECTION ASSESSMENTS WITH AN EMPHASIS ON PALEONTOLOGICAL COLLECTIONS

Kathy A. Hollis,[1] Dena M. Smith,[2] and Christina R. Spence[2]

[1]*University of Colorado Museum of Natural History, 265 UCB, Boulder, CO 80309-0265, current contact information: Department of Paleobiology, Smithsonian Institution, PO Box 37012, MRC 121 Washington, DC 20013-7012; hollisk@si.edu*

[2]*University of Colorado Museum of Natural History, 265 UCB, Boulder, CO 80309-0265*

Abstract.—Designing and completing a collection assessment can be a daunting, yet important exercise for collections managers and registrars. There are several excellent collection assessment case studies reported in the literature, but there is little guidance on how to begin and to customize an assessment for a collection, especially those with unusual space needs, such as paleontological collections. Outlined here is a guide for how to begin the process of conducting a collection assessment, including: preplanning; customizing categories, units, and rankings; quantifying space needs; data collection; data analysis; and mapping resulting needs.

Introduction

Collection assessments are essential for understanding a collection's strengths and weaknesses and allow one to focus resources on areas that need the most attention. Taking a quantitative approach to understanding the state of a collection is imperative to plan for future collection growth and configure collection storage space most efficiently. Having qualitative data about the health of a collection also is useful when writing grant proposals for collection improvement or for collection-based research projects.

Case studies of quantitative collection assessments have been well-established in the collections management literature (McGinley 1993; Williams et al. 1996; Hughes et al. 2000; Moser et al. 2000; Simmons and Muñoz-Saba 2003; Adrain et al. 2006; Favret et al. 2007; Camacho and Burneo 2009) and each study uses slightly different statistical approaches to identify and quantify collection health. In general, quantitative collection assessment methods involve using a categorical coding system to rank how well the collection meets predetermined collection standards. These ranks are assigned to a ''profiling unit''—a discrete unit of collection material (e.g., a drawer, cabinet, isle) to be assigned a single grade according to its state of health (Moser et al. 2000). Some studies use the ranks to calculate a health index (McGinley 1993); other studies map rank data onto collection layouts using a color-coding system (Favret et al. 2007).

When designing a collection assessment, it is important to customize data collecting methods to produce clear and meaningful results that are of greatest utility to you. Due to the variety of collections found within natural history museums and the unique needs of each, the health categories and ranking schemes used to capture collection health in one kind of collection are not the best way to profile health in another. For example, paleontology collections do not face the same conservation and pest issues that zoology collections deal with simply because skin, hair, and tissues are more sensitive to deteriorative agents than rock or permineralized bone. Thus, although it might make sense to use a health category of ''pest damage'' for specimens in a zoology collection assessment, it could be appropriate to omit it from a paleontology collection assessment.

Collection Forum 2010; 24(1–2):72–79

Another issue to consider when designing a collection assessment is how to assign an appropriate ranking scheme to adequately capture the range of health per health category. The number of numerical ranks per health category affect the way the health data are distributed and can make data interpretation difficult. For example, if the category of "conservation status" has two rank levels (i.e., rank of 1 means specimen is unstable and 2 means specimen is stable), one is unable to determine which unstable specimens need immediate conservation versus those that should be conserved soon, making prioritizing difficult. Alternatively, using 10 rank levels to score "conservation status" can introduce too much data and can muddle decisions concerning which specimens are priorities to conserve. In other words, the profiling unit used in an assessment will affect the resolution of the data being interpreted.

When using collections health data to create a tailored collections improvement plan, it might be easier to think of the collection as the number of specimens to be conserved, or the number of drawers, or the number of cabinets—or all three. Having a thorough understanding of what health information should be captured and at what resolution is an essential part of designing collection assessment procedures.

Within paleontological collections, understanding space requirements and potential space for future growth is of particular concern. Fossils often have highly variable sizes and shapes, especially when left unprepared out of the rock in which they were imbedded. Storing fossils with irregular dimensions in a cabinet can result in several unused cubic feet of storage space. However, arranging fossil storage with the primary goal of saving space is impractical for the way collections are used—fossils often are organized by geological time and formation in which the fossil was found. Even taxonomic organization cannot accommodate for size and shape inconsistencies due to preparation or preservation.

In this report, we outline a process that can be used for designing an effective quantitative collection assessment appropriate for the individualized needs of a collection. How to choose appropriate categorical variables to gather useful, interpretable data that can help improve the health of a collection will be reviewed. In addition, general procedures appropriate for assessing storage space specific to paleontological collections will be discussed.

Questions to Ask Before Designing Your Assessment

Time spent planning is important to any collections project, and will help identify problems and troubleshoot them in advance. Having answers to the following questions will allow you to develop your plan for data collection and will insure that the data you collect are complete and consistent.

1. Why do you need to profile the health of your collection? Was the collection previously neglected? Are you planning to apply for research or collections improvement grants? Are you expecting a large-volume acquisition?
2. What is the time frame that you have to complete the assessment? Will this project be contingent on funding deadlines?
3. Who are the people who will be working on this project? How many hours can be dedicated to data collection for the assessment?
4. How thorough an assessment can you complete given your available financial or human resources? Is there one portion of the collection or one aspect on which you should focus or omit altogether? Can you collect more data than is immediately needed in preparation for future needs?

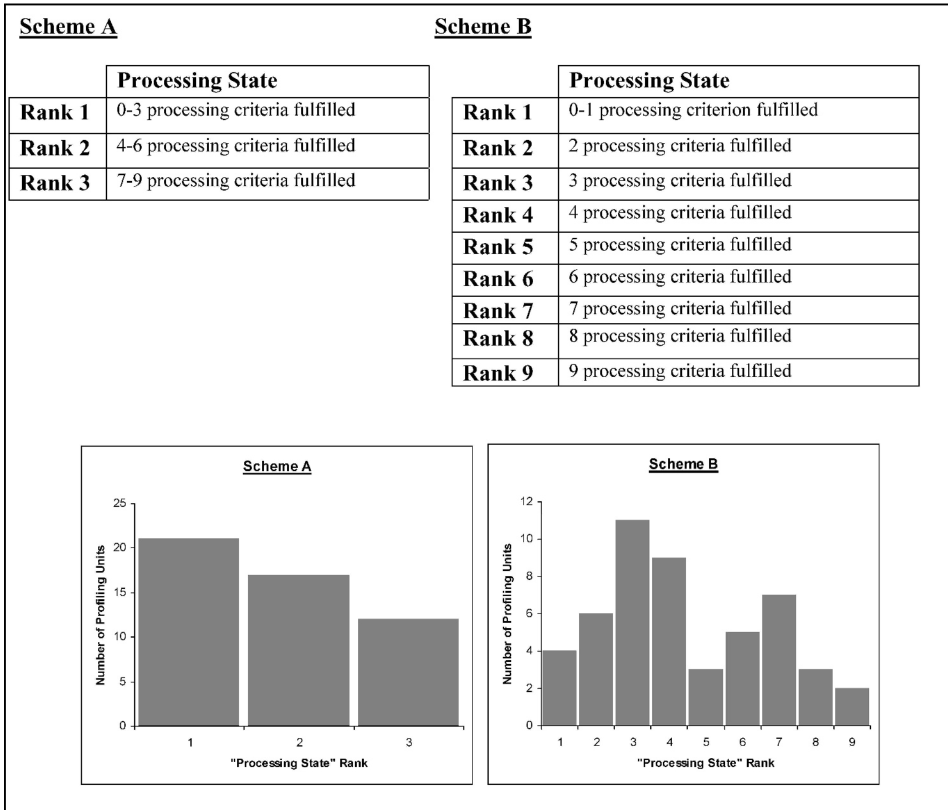

Figure 1. Scheme A and Scheme B illustrate two different ranking schemes to describe how completely a profiling unit has been processed. The same hypothetical data were used for both schemes. Processing criteria might include proper housing materials, complete catalog record, complete database record, database record includes digital images of specimens, etc. Scheme A is best for capturing a broad snapshot of collection health; Scheme B is best for capturing details of where specific collection health issues are present.

Choosing Health Categories, Ranking schemes, and Profiling Units

The health categories you choose to evaluate, the ranking scheme you use for each category, and the profiling unit you measured for each category will shape the outcome of your collection assessment. Categories should capture best the state of the collection according to your predetermined standards of curation (e.g., types of specimen tray used, label, completeness of documentation). The categories used within Moser et al. (2000)—conservation status, processing state, storage containers, arrangement, specimen identification, and inventory—cover the basic variables of collection health, but you can add other categories to capture additional health qualities that you deem necessary to measure.

Each category should have a ranking scheme in which the gradation between "best" and "worst" conditions can be reflected by a numerical value. Figure 1 shows an example of two different ranking schemes used to describe the health category "Processing State" and how these two schemes might describe the same hypothetical data. When you are determining how many ranks per each category, consider the differences between *best* and *worst* and how many corrections would have to be made to change a *worst* profiling unit

to *best*. The number of conditions that must be met to have a *best* state can be your highest rank. For example, if there are five conditions to be met, profiling units that meet all five conditions will get a rank of 5. If only four conditions are met, that unit will be ranked 4, three conditions met will receive a 3, etc. If you decide there are too many conditions to have an optimal state for a health category, it might be more beneficial to split one category into two or more categories so you can identify which condition (e.g., numbering, labeling) needs to be addressed.

Depending on how you plan to use your data, data interpretation might be easier if all categories have the same number of ranks. For example, conservation status, processing state, and storage containers all might have three possible ranks that reflect *best*, *moderate*, and *poor* states (e.g., ranks 3, 2, and 1 respectively). This type of approach is most useful when you want to take a broad, overall look at the health of multiple categories at the same time. At a glance, you will know that all profiling units with a rank of 3 currently are at their optimal state. Alternatively, using the same number of ranks for each health category might not adequately capture the state of each health category. Three ranks might be necessary to measure conservation status, but you might want to look into the state of processing in more detail and use five ranks instead of three.

Whatever approach you choose, each health category and its respective ranking scheme should be concisely defined in writing. These definitions can be referred to as needed during data collection to avoid ranking errors and inconsistencies, particularly if several different people work on scoring each profiling unit.

Once you know the kind of data you need, choose a profiling unit appropriate to your assessment. Moser et al. (2000) states that "profiling units are the discrete subsets of the collection that are evaluated according to the scoring categories …." Your profiling unit should be small enough to get the resolution you need, but large enough to prevent data collection from becoming overly cumbersome. If you want to analyze your data in multiple ways (e.g., by cabinet, formation, or taxon), make sure each group can be extracted from the profiling unit.

Capturing Space Requirements for Paleontological Specimens

Paleontological collections are susceptible to deterioration from physical forces as defined by Waller (1994). Common storage issues include specimen lots where specimens are piled together in a box, or specimen boxes stacked upon each other in drawers. In addition, specimens often are stored in drawers that cannot accommodate their height, which causes the specimens to scrape the drawer above them every time the drawer is opened or closed. Issues such as these arise when a collection is short on available space or when specimens were incorrectly curated in boxes that are too small. In either scenario, collection space usually is lost in order to mitigate the problem.

Storage space needs should be assessed by evaluating each profiling unit by how much expansion space is needed to accommodate stacked or cramped fossils compared to how much space is available in profiling units that are empty or not filled to capacity. We found the easiest way to accomplish this was to make *expansion requirements* a health category. Our ranking scheme for expansion requirements was as follows: Rank 1—the profiling unit must be expanded to over half its current volume; Rank 2—the profiling unit must be expanded to less than half its current volume, and Rank 3—there is sufficient space for the profiling unit's contents. To measure available space within the collection, a similar ranking scheme was developed to measure *available space* as a health category: Rank 1—No space available, Rank 2—half of the volume of the profiling unit

Table 1. Ranking schemes for *expansion requirements* and *available space*. In a sense, the two categories are inverses of each other. Making each a separate health category allows for easy data comparison and analysis.

Rank	Expansion requirements	Available space
1	Over half the of profiling unit is needed for expansion	No space in profiling unit is available for expansion
2	One half or less of the of profiling unit is needed for expansion	One half or less than the profiling unit is available for expansion
3	No expansion space needed	Over half of the profiling unit is available for expansion

is available for expansion; and Rank 3—Profiling unit almost or completely empty. If you have a cabinet with unused and available drawer slots (and you have drawers on hand to fill the slots), be sure to treat those slots as profiling units when recording *space available* data. Table 1 compares the *expansion requirements* and *available space* ranking schemes. Using these ranks, the available and overcrowded space in your collection can be tallied, averaged, and illustrated on a collection layout. Your space surplus or deficit can be calculated by subtracting the values for space needed from the values for space available.

Data Collection

Consistency in data collection is crucial, especially if multiple people are assigning ranks to profiling units. Be sure to take enough time to train all workers to ensure that their work is consistent. Also, as workers become practiced in quickly assigning ranks to profiling units, there is a risk of inconsistency within an individual worker's data set. To avoid the data being biased within or between workers, the definitions of each health category's rank must be specific and detailed. A worker should not have to guess about whether a drawer of specimens should be ranked poor or fair. We recommend building in quality control checkpoints during the data collection phase. The person overseeing the assessment should spot-check the rankings of one profiling unit for each worker when data collection is 25%, 50%, 75%, and 100% complete. Ideally, the way profiling units were ranked should not be affected by who assigned the rank or by when the rank was assigned.

Your data should be digitized in spreadsheet form for ease of manipulation and analysis. Your data sheet can be as simple or as complex as you need. An Excel spreadsheet is sufficient, but given your needs and resources you might want to put your data in a relational format such as an Access database or add fields to your existing specimen database. Table 2 shows a sample data table and Figure 2 shows a map of the data for Conservation Status based on the sample data table. The columns list each category and the rows list each unit ranked. It might be helpful to include a column for "notes" where you can include any information not captured in the health categories, such as the presence of old loan slips or missing specimens that must be addressed by the collections manager.

There are a few options for how workers can input their data. The most low-tech option is to give each worker a hard copy of a blank spreadsheet to fill in as they are working. Later, they can enter their data into a master digital file. If you have a small workforce, it might be more efficient to enter data directly on a laptop that a worker can take with them as he or she moves through the collection. The most high-tech option is to have workers use speech recognition software, Bluetooth headsets, and a nearby laptop

Table 2. Sample data table for three cabinets with three drawers each.

Profiling unit	Conservation status	Processing state	Storage containers	Expansion requirements	Available space	Notes
Cabinet 1						
Drawer 1A	1	3	2	2	1	2 expired loan slips
Drawer 1B	2	3	1	1	1	
Drawer 1C	3	2	1	3	2	Contains field notes
mean	***2***	***2.67***	***1.33***	***2***	***1.33***	
Cabinet 2						
Drawer 2A	1	2	2	1	1	
Drawer 2B	1	2	2	1	1	
Drawer 2C	1	3	1	3	3	
mean	***1***	***2.33***	***1.67***	***1.67***	***1.67***	
Cabinet 3						
Drawer 3A	3	1	3	2	1	
Drawer 3B	3	1	3	2	1	One missing specimen
Drawer 3C	3	3	3	2	1	
mean	***3***	***1.67***	***3***	***2***	***1***	
Collection mean	***2***	***2.21***	***1.99***	***1.89***	***1.33***	

to enter data simply by speaking. This greatly can improve the speed at which data can be collected if the data being collected are primarily numerical, which are very well-suited for speech recognition software.

Data Analysis and Interpretation

To draw meaningful conclusions from your data, the average scores for each health category must be calculated. If your profiling unit is a specimen drawer, we recommend calculating averages for (1) the entire cabinet, (2) each aisle of cabinets, and (3) the entire collection. Keep in mind how much detail you need versus how broad a picture you need.

We recommend using similar methods to Favret et al. (2007) to map your data and related calculations on your collection layouts. That way, you can quickly pinpoint where the problem areas are in your collection. You might find that a problem area for one health category is different from a problem area in another. If your data collection schemes were well planned, you should be able to answer any questions about your collection's health by doing simple calculations, creating histograms, and generating collection data maps to compare and analyze health.

When using your assessment data to plan for collection improvement, think about how each health category can affect and overlap with one another. Prioritize problems to address based on (1) the overall goals of your assessment, (2) your available resources to mediate issues, and (3) a logical, practical order in which to mediate issues.

Concluding Remarks

To conduct an effective collection assessment it is important to spend some time a priori designing your data collection protocols. This preplanning phase will facilitate timely and accurate data collection, analysis, and eventual prioritization and planning. This process is especially beneficial for those who manage large collections with varied needs and can help to clearly define needs and goals. Using this guide for getting started

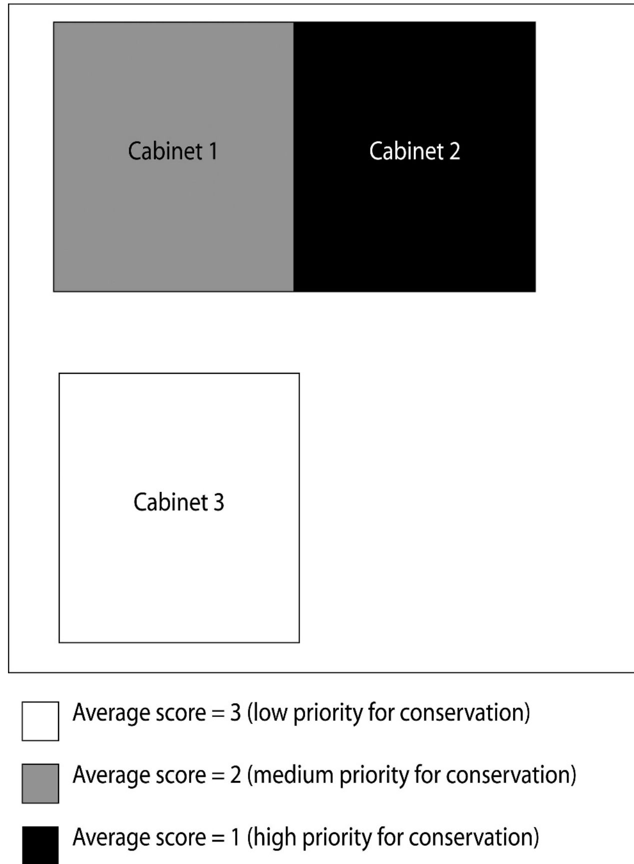

Figure 2. Hypothetical collection map based on *Conservation Status* data from Table 2. According to the data, the cabinet is the highest priority for conservation needs is Cabinet 2.

and other published case studies should facilitate collection assessment no matter what the collection or needs may be.

Acknowledgments

We thank the reviewers, Jessica Cundiff and Talia Karim, and Managing Editor Susan Butts for their helpful comments and editorial suggestions.

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THE UTILITY OF CHICKEN BROTH IN THE PREPARATION OF SKELETONS FROM FRESH AND FLUID-PRESERVED VERTEBRATE SPECIMENS

Kirsten E. Nicholson and Dana M. Smith

Museum of Cultural and Natural History, and Department of Biology, 103 Rowe Hall, Central Michigan University, Mount Pleasant, MI 48859

Abstract.—Vertebrate osteological (skeletal) collections are essential for both research and educational purposes. Fresh specimens can be skeletonized following several methods, but sometimes it is desirable to produce skeletons from previously preserved museum specimens. Several methods have been proposed for producing dermestid-cleaned skeletons from fluid-preserved specimens, and here we report our results using an additional method. We investigated the effect of using a chicken broth pretreatment that proved to be easy and cheap, worked successfully with preserved material, reduced the cleaning time of both fresh and chemically preserved fish, and also facilitated dermestid cleaning of a maggot-contaminated specimen.

Introduction

Osteological (skeletal) material forms an essential component of museum-based vertebrate collections for both research and educational purposes. Species often are defined by important skeletal features that are not visible externally (e.g., Amaral et al. 2009; Reding et al. 2009; White et al. 2009). Geographic variation within species may be detected by comparing skeletal features, and is sometimes determined to be of great evolutionary importance (e.g., Gould and Johnston 1972). In addition, instructing students of biology at any age (K–12 and university) is greatly facilitated by presenting osteological material that depicts the key morphological features of vertebrates.

Specimens of some taxonomic groups (most notably, fish, amphibians, and reptiles) routinely are fixed in formalin and stored in ethanol (Hildebrand 1968; Hangay and Dingley 1985; Hawks 1999; Simmons 2002). In some cases, bird or mammal specimens are fixed and/or preserved in fluids for a variety of reasons (e.g., materials for standard preparation are unavailable when a deceased, but unique, specimen was encountered). The conversion of fluid-preserved specimens to dry osteological preparations sometimes is desirable for comparative research or educational purposes, and various methods have been proposed to accomplish this.

The use of dermestid beetles (Dermestidae, *Dermestes* spp.) has been long-employed by museums for preparing skeletal material, and is a relatively fast, easy, and inexpensive method. Beetle colonies are fairly easy to establish (Grayson and Maser 1978; Hangay and Dingley 1985; Hildebrand 1968), although they must be monitored carefully, and set up in areas completely enclosed and separated from collection storage areas to pre-empt their escape and damage to existing collections (Hangay and Dingley 1985). Humidity and temperature must be controlled such that the beetles remain active while mold infestation of the colony is prevented (Hildebrand 1968). Accommodating these conditions is reasonably easy; however, specimens that have been fixed or preserved in chemicals will not be consumed by dermestids without prior processing. Previously suggested procedures for encouraging dermestids to accept chemically treated specimens include water baths, to leach out the preservatives, followed by a variety of pre-treatments, such as the addition of animal grease (De La Torre 1951), or dipping in cod liver oil (Hooper 1956; Hildebrand 1968), or immersion in a dilute blood solution (Gritis

and Brunner 1990), or beef broth (Hildebrand 1968; Dirrigl et al. 1993). These treatments vary in their effectiveness and ease of use, and any method that is inexpensive, reasonably fast, and easy to employ, is preferable when attempting to develop osteological collections from preserved material.

We conducted a series of experiments to investigate the efficacy of presoaking specimens destined for dermestid cleaning in chicken broth. Specifically, we addressed the following questions: (1) Will dermestid larvae consume previously preserved vertebrate specimens soaked in water followed by soaking in chicken broth? (2) Will dermestids consume fresh specimens (not previously treated with any chemicals) immersed in chicken broth as fast as, or faster than, fresh specimens? (3) Are there taxon-specific differences in soft tissue consumption by dermestids? These last two questions were stimulated, at least in part, by a recent study (Bemis et al. 2004) that investigated methods for preparing fish skeletons with dermestids. Bemis et al. (2004) described a method for preparing fish skeletons using a drying process followed by dermestid cleaning, which can take weeks or months to complete, depending on a variety of factors such as humidity, size of colony or specimens, etc. We therefore investigated whether pretreatment with chicken broth might also facilitate the preparation of fish skeletons, both fresh and preserved, by dermestids.

Methods

Preserved specimens were selected from the vertebrate collection of Central Michigan University's Museum of Cultural and Natural History. Specimens were selected according to the following criteria: they represented diverse vertebrate taxonomic categories (fish, amphibians, reptiles, and birds) that had been fixed in formalin and stored in ethanol, and were species that were numerous in the collection, and for which we lacked skeletal material (Alewife, *Alosa pseudoharengus* MCNH F-1556; Black-capped Chickadee, *Poecile atricapillus* MCNH B73-54; Eastern Milksnake, *Lampropeltis triangulum triangulum* MCNH R-34; Traill's Flycatcher, *Empidonax traillii* MCNH B73-14; and Western Leopard Frog, *Lithobates pipiens* MCNH A-398a). There were no fluid-preserved mammals for which we had a plentiful collection, so we obtained the carcasses of laboratory mice (*Mus musculus*) used for other experiments that had been euthanized in the Neuroscience Department at Central Michigan University. These mice were not fixed in formalin, but were stored in 95% ethanol for 2 months prior to the initiation of this experiment.

We compared the dermestid preparation of fluid-preserved specimens to select "fresh" specimens (defined as never having been chemically treated, but frozen prior to the initiation of this experiment) to serve as controls. We used specimens representing the same taxonomic categories as above (fish, amphibians, reptiles, and birds), although identical species were not always available. If multiple individuals of the same species were not available, we selected alternatives that were about the same size as the preserved specimens to control for size-related differences in dermestid processing. Four "fresh" specimens (as defined above) also were immersed in chicken broth to compare dermestid cleaning time to the controls and other experimental treatments.

Previously preserved specimens (one individual from each of the above categories) were soaked in a water bath at room temperature for 2 weeks, changing the water every 1–2 days. A broth of chicken bouillon was prepared from crystals of Wyler's® Instant Bouillon purchased at a local grocery store and dissolved in warm water (1 teaspoon of crystals/quart). (Bouillon ingredients include salt, sugar, corn maltodextrin, hydrolyzed corn protein, monosodium glutamate, chicken fat, onion powder, cooked chicken

powder, turmeric, natural chicken flavor, gelatin, autolyzed yeast extract, disodium inosinate and disodium guanylate, garlic powder, corn syrup solids, natural flavors, celery seed, modified corn starch, hydrolyzed soy protein, soybean oil, partially hydrogenated soybean oils, tricalcium phosphate, tertiary butylhydroquinone [preservative], artificial flavor, alpha tocopherol [antioxidant], butylated hydroxyanisole [preservative], propyl gallate, citric acid, and butter oil. The product also contains soybeans and milk powder, and was processed on equipment that also processes wheat, soybeans, milk, and eggs.) Each specimen was placed in a plastic container with a tight-fitting lid and rubber gasket. Broth completely covered the specimen, and was refreshed every 3–4 days (placement of the container on a rocker table to keep the broth moving gently, but constantly, slowed stagnation of the broth at room temperature) for a total soak time of 1–2 weeks, depending on the size of the specimen. After soaking, specimens were transferred to the dermestid colony for cleaning. The four fresh specimens that were immersed in chicken broth also were subjected to the above procedure, but were not soaked in water beforehand.

Prior to each test, all food items were removed from the dermestid colony for 3–4 days. Each specimen was then placed within the colony individually, and checked approximately every 2 hours throughout the day to record progress. We allowed cleaning to continue until the entire specimen was cleaned, and then recorded the total time required for cleaning and removed the specimen. The next specimen was then placed in the colony after waiting 3–4 days, and the process continued until all tests had been conducted. All tests were conducted in June of 2009.

Results

Previously preserved amphibian, reptilian, avian, and mammalian specimens were successfully and completely cleaned, with no ill affects to the dermestids, within a few hours of introduction to the dermestid colony (2 to 36 hours, depending on the size of the specimen; see Table 1). In terms of the removal of tissue, there appeared to be no difference in the quality of the cleaning by dermestids of previously preserved specimens treated with chicken broth compared to fresh specimens. There were no signs of damage to the skeletons as a result of chicken broth immersion and dermestid cleaning.

Fresh specimens immersed in chicken broth were cleaned as rapidly or more rapidly than those not treated with chicken broth (fish specimens in particular, see below). Most specimens, depending on size, were cleaned within only a few hours. Fish specimens (both previously preserved and fresh) were cleaned much faster after pretreatment with chicken broth compared to those that were not pretreated. Previously preserved fish soaked in water and immersed in chicken broth were completely cleaned by dermestids within 4 to 6 hours. Fresh specimens pretreated in chicken broth were completely cleaned in the same or less time than it took to clean the preserved specimens, and in the case of the fish specimen, in less than 24 hours.

Discussion

Osteological material is an important resource with which to demonstrate key anatomical differences between vertebrates. There are important features of the skeleton that can be examined by researchers to reveal geographic variation within species or differences between species that cannot be observed externally. The development of osteological collections is an important aspect of vertebrate museum collections, and contributes to both educational and research initiatives.

Table 1. Specimens used to test the efficacy of chicken broth immersion on dermestid cleaning. Specimens without catalog numbers were not accessioned into the collection. All preserved specimens were soaked in water for 2 weeks at room temperature, with water changed every 1–2 days. Specimens (both fresh and preserved) immersed in chicken broth were soaked at room temperature for 1 week in all cases, except for the snakes, which were immersed for two weeks, changing the broth every 3–4 days.

Specimen	Size Prior to Skinning	Initial State	Soaked in Chicken Broth?	Dermestid Cleaning Time
Alewife *Alosa pseudoharengus* (MCNH F-1556)	13 cm	Formalin fixed & Ethanol Preserved	Yes	4-6 hrs
Black-capped Chickadee *Poecile atricapillus* (MCNH B73-54)	9.5 cm	Formalin fixed & Ethanol Preserved	Yes	2-4 hrs
Eastern Milksnake *Lampropeltis triangulum* (MCNH R-34)	53 cm	Formalin fixed & Ethanol Preserved	Yes	24-36 hrs
Traill's Flycatcher *Empidonax traillii* (MCNH B73-14)	15.5 cm	Formalin fixed & Ethanol Preserved	Yes	6-12 hrs
Western Leopard Frog *Lithobates pipiens* (MCNH A-398a)	11 cm	Formalin fixed & Ethanol Preserved	Yes	4-6 hrs
Domesticated Mouse *Mus musculus*	8 cm	Ethanol Fixed & Preserved	Yes	2-4 hrs
Yellow Perch *Perca flavescens*	15 cm	Fresh	No	2-4 hrs
Black-capped Chickadee *Poecile atricapillus*	9.5 cm	Fresh	No	2-4 hrs
Northern Watersnake *Nerodia sipedon*	47 cm	Fresh	No	12-24 hrs
Cedar Waxwing *Bombycilla cedrorum*	18 cm	Fresh	No	4-6 hrs
Western Leopard Frog *Lithobates pipiens*	12.5 cm	Fresh	No	4-6 hrs
Domesticated Mouse *Mus musculus*	8 cm	Fresh	No	2-4 hrs
Yellow Perch *Perca flavescens*	15 cm	Fresh	Yes	2 hrs
Black-capped Chickadee *Poecile atricapillus*	9.5 cm	Fresh	Yes	2 hrs
Cedar Waxwing *Bombycilla cedrorum*	18 cm	Fresh	Yes	2-4 hrs
Domesticated Mouse *Mus musculus*	8 cm	Fresh	Yes	2 hrs

Our study shows that the dermestid preparation of preserved vertebrate specimens was notably enhanced by immersion in chicken broth. Some "fresh" (i.e., not previously chemically fixed or preserved) specimens were cleaned faster after soaking in chicken broth compared to those not pretreated in this way. Previous studies (Bemis et al. 2004,

and references within) commented on the difficulty with which fish skeletons are prepared by dermestids, but we found that fish specimens soaked in chicken broth were cleaned rapidly without the need to dehydrate specimens beforehand. We were even able to successfully clean a maggot-infected specimen—a Cooper's Hawk, *Accipiter cooperii*, MCNH 2008-5—that the dermestids would not touch (even after freezing and removing the maggots) after pretreatment with chicken broth. Immersion in chicken broth, particularly after soaking in water to leach out preservatives, appears to make the tissues more palatable to dermestids without any adverse effects to the skeleton or the dermestids themselves.

Several methods previously have been proposed for producing dry osteological material from fluid fixed and/or preserved specimens, and we now add immersion in chicken bouillon to this list. Chicken broth immersion is a useful method for facilitating dermestid cleaning of preserved specimens, and even of maggot-contaminated specimens. We are aware that other museums have used chicken broth pretreatment in a similar fashion (Nicholson pers. comm.), but are not aware of any published account describing or experimenting with its use. The addition of more replicates for each treatment, investigating the effects of such variables as length of time in fixative and preservative on the efficacy of dermestid cleaning, length of time required to soak in water to leach out the chemicals, and minimal length of time required to soak specimens in the chicken broth, all clearly would be of value. Nevertheless, this study shows that chicken broth immersion can successfully assist in the production of osteological material from both fresh and preserved vertebrate specimens.

Acknowledgments

Financial support for both authors was provided by the Museum of Cultural and Natural History (D.M.S. entirely, K.E.N. partially) and the Department of Biology (K.E.N. partially), both at Central Michigan University. We would like to thank Angela Dassow (University of Wisconsin Zoological Museum), John Friel (Cornell University Museum of Vertebrates), Tomas Labedz (University of Nebraska State Museum), Dirk Neumann (Zoologishe Staatssammlung München), and Greg Schneider (Museum of Zoology, University of Michigan), for advice and comments, although those presented here are strictly our own. Sincere thanks also go to Frank Dirrigl, Maureen Flannery, Jeremy Gibson-Brown, John E. Simmons, and one anonymous reviewer for editorial comments and suggestions on previous versions of the manuscript

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