AN INVESTIGATION OF THE METHODS OF PREPARING AND MOUNTING INSECTS FOR PERMANENT PRESERVATION*

FRED O. DODD

I. INTRODUCTORY

Insect collections are valuable for reference and research, for teaching and for exhibition. There can be no distinct groupings into these classes except by personal opinion, the differentiation between reference and research being perhaps the most difficult. Certain institutions set apart in special collections a limited number of specimens that are of special significance and the bulk of their specimens is included in a larger collection. These smaller collections are often held inaccessible to persons not actually engaged in their preparation while the larger ones are available for study or reference.

Specimens are exhibited for various reasons and such groups of individuals comprise an exhibit collection. It may contain a varying number of specimens depending upon its purpose.

The collection to be used in teaching varies, too. It is often quite incomplete when compared with a collection of the first group because of the fact that representative specimens are most commonly used in teaching.

Regardless of its ultimate use a collection should be properly prepared, mounted and labeled if it is to render its greatest usefulness. The increase in interest in Entomology has brought a decided increase in collection and many private collections have been and are being built up. Many specimens in these collections would, if properly mounted, throw light upon some entomological problems that are quite insufficiently solved now. It is with this in mind that this paper has been prepared and it is the hope of the author that it may be a step toward the betterment of general entomological technique. An effort has been made to bring together practical methods of preparing and mounting insects with special reference to the permanency of the preservation.

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II. METHODS OF KILLING INSECTS.

In killing insects the agents employed must act quickly. This is necessary to prevent specimens damaging themselves or others in the same container in their efforts to escape confinement. The manner of killing, therefore, is closely related to the methods of mounting and the purpose for which specimens may subsequently be used. Many insects must be kept free from surface moisture if proper mounting methods are to be employed and killing agents that cause hardening and contraction of the muscles or undue propulsion of cervical organs should not be used (see p. 301). With special groups certain added precautions should be taken and collectors should make an effort to learn the use for which their specimens are intended in order that they can supply material best suited for that particular purpose. Commercial concerns may find it advisable to furnish their collectors with materials and instruction and to refuse to accept specimens in other than the specified condition while individuals who mount their own material find practically no problem with killing methods.

Cyanide is the most widely used killing agent. Salts of potassium, sodium and calcium are available in rock or granulated form. The salts of calcium are not stable and are not practical for use in cyanide bottles and potassium cyanide is considered superior to sodium for killing insects. Cyanide bottles are prepared in various ways, three of the most practical of which will be discussed here: (1) place a liberal amount of cyanide in the bottom of a glass vial or container of convenient size and cover with a filler composed of dry sawdust, cotton or plaster of Paris. Then add a layer of freshly mixed plaster of Paris paste (plaster of Paris and water) and stand aside uncorked until dry. Before using the sides should be scraped free from paste; (2) a second method is similar to the first. A disc of porous sheet cork is substituted for the plaster of Paris paste and is superior to the first method in ease of renewal and cleanliness. The plaster of Paris has a tendency to rub off when it becomes damp; (3) the charged cork method is superior to both of the above if the cork used is large enough to admit the use of a sufficient amount of cyanide. The cyanide is placed in a small vial or "cell" as it is called and

inserted into a hole bored into the cork. The gas penetrates the cotton used to retain the cyanide. Bottles so made have distinct advantages: (1) specimens need not be transferred to other containers after death—the cork can be traded for that of another vial or bottle and (2) the bottles can be cleaned more easily after a period of collecting.

The disadvantages of cyanide as a killing agent can in most cases be traced to negligence on the part of the collector. Salts of cyanide are hygroscopic and the moisture that collects in the bottle is quite objectionable. This condensation can be prevented by use of blotting paper or other absorbent paper. This paper can be renewed when necessary. The yellow colors of some Hymenoptera, Lepidoptera, and Diptera turn reddish when exposed to cyanide too long. Such specimens should be removed soon after death. Specimens often become dirty when killed in cyanide bottles. Care in excluding dirt particles or dirty specimens removes this objection almost completely altho the careful collector finds the time used in cleaning his collecting bottles well spent. Lepidoptera and bees should not be killed in the same bottle with other insects as the scales from the wings of the former are easily dislodged and are extremely hard to remove from other specimens and pollen from bees raises a similar objection. In no case should larvae be killed in a cyanide bottle with other forms.

Hydrocyanic acid gas which is the active killing agent of cyanides is one of the most deadly gases known. Its action upon insects is not definitely known, but the fact that forms that respire rapidly are killed more quickly than other forms whose rate of respiration is much slower indicates that the effect is upon the respiratory system, probably producing both suffocation and paralysis. There is little danger from cyanide if handled properly although small amounts produce quick death to man and small animals and if left exposed to the atmosphere in a closed room may generate enough gas to produce death when the room is again entered. Too great precautions cannot be taken in handling it.

Alcohol has been used as a killing agent. Insects are dropped into weaker alcohol—usually 50 per cent— and after death the specimens are transferred to higher percentages. Preservation is accomplished at 70 per cent altho many workers prefer to use 80-95 per cent. Ethyl alcohol is the most satisfactory form for use as a killing agent, but it is hard to procure and cannot be carried about with safety even though it is for scientific purposes. When

collection is being done for commercial concerns good alcohol so treated as to be unfit for human consumption may be supplied, although there are few instances where alcohol is a very satisfactory killing agent. Its powers of pentration are too weak and because of its power as a dehydrating agent water is drawn from the body of the specimen faster than it is replaced by alcohol and shrinking from unequal pressure results. The outer tissues are hardened in this abnormal position and poorly shaped specimens are often obtained by killing in alcohol. Jackson ('07) has obtained satisfactory results with killing Thysanoptera in boiling absolute alcohol although heat is probably a great factor as we shall see in the next paragraph.

Hot water has a very definite place as a killing agent. It is especially good for soft-bodied forms and is probably the best known means of killing larvae and pupae. Kelsheimer ('28) lists the following advantages: "(1) it kills bacteria and prevents discoloration; (2) it coagulates proteins; (3) it straightens and preserves body form; and (4) leaf feeding forms lose less green coloring matter when later preserved in alcohol." The water should not be boiling vigorously. Tothill ('19) recommends that water at a temperature of 80°-90° C. be poured over the specimens. Equally good results may be obtained by pouring the water on the specimens or dropping them into it. They should be left in the water from one to five minutes depending upon the species. The time for a particular species must be determined by each collector. Specimens sometimes shrink if removed from the water too quickly or air spaces occur under the cuticle if heated too long or too vigorously, but these disadvantages can be overcome by careful work. Specimens that have a tendency to shrink when dropped into hot water may be killed by placing them in cold water and gradually heating until death occurs. This practice is not advisable because death does not occur suddenly enough especially for histological purposes.

Etheric oils—chiefly ether, chloroform and benzene—are often used as killing agents. They kill rapidly and specimens remain relatively cleaner than with other methods although the muscles become drawn and hardened which renders setting or spreading of wings quite difficult in most cases. The heads of insects such as Diptera and Hymenoptera often drop off because of undue propulsion of some organs in the cervical region. Specimens may revive after apparent death and care must be taken

to prevent such occurrences. Some workers prefer to inject ether or chloroform into the thorax of large moths and butterflies and the most practical use of etheric oils is probably the quieting of larger forms to prevent injury in their efforts to escape from the cyanide bottle before death.

III. FIXATION OF INSECT SPECIMENS.

"Fixation usually infers two things: rapid killing of the organism to retain its form and hardening to such a degree that it will withstand the effects of reagents with which it may subsequently be treated without change in form." (Lee, 1928, page 2.) After fixation with some process known as a fixing agent the specimen should in most cases be washed before final preservation.

Various combinations of alcohol and other substances have been used as fixing agents. One of the most widely used and best recommended fixers is Bouins Picro Formol. It is made under the formula—

Picric acid (Saturated Aqueous Solution)....	75 parts
Formalin	25 parts
Glacial acetic acid.....	5 parts

This must be washed out with 70 per cent alcohol after fixation for 12-24 hours, the optimum being about 18. Warm alcohol hastens the washing process considerably. According to Lee ('28), 1 part of picric acid dissolves in 86 parts of water at 15°C. It is more soluble in warm water.

Another picric acid combination known as picrosulfuric acid is recommended as a good fixer where it is necessary to penetrate chitin. Lee recommends its use for Orthoptera and gives the formula—

Distilled water	100 vol.
Sulfuric acid	2 vol.
Picric acid	Saturation

The usual procedure is to dilute this stock solution with three times its volume of water. This must be washed out with warm 70 per cent alcohol.

Ble's solution is recommended especially for embryological purposes and has the advantage over other fixers that the specimens need not be transferred to other preservatives. It is not stable and should not be used as a fixer after two weeks, altho it remains perfectly good as a preservative after that time. A stock solution

of 90 cc. 70 per cent alcohol and 3 cc. of glacial acetic acid should be made up to each 93 cc. of which 7 cc. of formalin should be added when one is ready to use it. The author has found this very good for the preservation of eggs, larvae and pupae of honey bees and some other soft-bodied forms.

Similar to, but stronger than Ble's solution, is one known as Carl's solution. Fixation is more rapid and overfixation begins after 24 hours. Specimens should be transferred to other preservatives. Washing is not necessary. The formula is—

Absolute alcohol	15 parts
Formalin	6 parts
Glacial acetic acid	2 parts
Distilled water	30 parts

Rawlin's solution is quite similar to the above two and is recommended as a good process by which to retain colors in stems, leaves and fruits of plants. The author has had little success with this fluid in fixing insects. The formula is—

Alcohol 70%	100 cc.
Formalin	2 cc.
Glacial acetic acid	5 cc.

Acetic alcohol is one of the most penetrating and quickly fixing reagents known and the two following formulae are recommended: Carnoy's Acetic Alcohol—

1.	Absolute alcohol	3 parts
	Glacial acetic acid	1 part
2.	Absolute alcohol	6 parts
	Glacial acetic acid	1 part
	Chloroform	3 parts

The addition of chloroform, according to Lee, hastens the reaction and is recommended where extremely rapid fixation is desired.

Kelsheimer ('28) has used uranium nitrate with promising results in preparation of egg masses of the European corn borer for the museum. He fixes for 2-5 days and transfers to alcohol. The formula is—

Uranium nitrate	1.5 gr.
Ethyl alcohol (95%)	90 cc.
Formalin	5 cc.
Glacial acetic acid	2.5 cc.
Glycerine	2.5 cc.
Copper chloride	5 gr.

Alcohol alone is recommended by Tillyard ('26) and Kelsheimer ('28). Tillyard also recommends Formalin 1 part to 10 parts of water, but classes both formalin and alcohol second to Ble's solution. Kelsheimer uses boiling 80 per cent alcohol as a fixative for white eggs to be preserved on the green leaves upon which they were deposited.

Kingsbury and Johannsen ('27) claim that 10 per cent formalin is a fair fixer satisfactory for gross study and museum preparation. They say that it is preferable to use formalin in connection with other chemicals. They also say that 95 per cent alcohol answers admirably for most histological study, fixing well, hardening and likewise dehydrating. In Lee ('28) we find an opposing view. Alcohol is said to be a fair dehydrating agent, but far less satisfactory as a fixer. Lee's contention is that no single substance or chemical fulfills all the requirements of a fixer and that alcohol is seriously lacking, especially for histological preparations.

Kelsheimer and others recommend that larger forms should be injected with formalin or some other fixing solution immediately after death or that death be produced in that manner. Very good results have been obtained in preservation of forms for study of internal anatomy in this way.

IV. Methods of Mounting Insect Specimens.

When the subject of mounting insect specimens is mentioned one thinks first of some use of pins as this is probably the most widely used method. There are several other methods, the discussion of which will follow in succeeding paragraphs. Many kinds of pins have been used by workers ranging from the short nickeled pins of German manufacture, to the black "Japanned" or "enameled" pins in use by workers of today. These earlier pins were nickled or tinned and were about the size of the ordinary dress pin. They were not very satisfactory because of a tendency to rust easily, and verdigris (see p. 323) was very common. They corroded when used in mounting practically all insects. Later workers used a longer pin and various sizes began to be used very soon. The efforts to eliminate verdigris brought about the manufacture of the japanned or enameled pin by a German concern. This was later taken up by American manufacturers with various claims for superiority of product. Pins of both German and American manufacture are available at supply houses at a nominal

cost and verdigris has been materially reduced by their use. American steel pins are also available, but they have a tendency to rust easily and are therefore not as satisfactory as japanned pins.

Very early, there was a tendency to use some form of what Tillyard ('26) calls double mounting. Various kinds of wire were used as substitutes for pins and a German concern produced a very fine pin known as the minutien-nadeln pin in two styles. One was made by pointing a straight piece of silver wire for entrance into both the insect and the secondary mounting structure and the other was known as the elbow pin. It was pointed on one end and bent so as to lie parallel to a larger pin passed through a coil in its opposite end. Such pins are in common usage by some modern workers for very minute insects.

Perhaps the most common method of mounting very minute insects is on points—triangular strips of paper or mica supported at their bases by No. 2 and No. 3 pins. There are many modifications of this method. Mica was perhaps first used because of its transparency. The specimens could be viewed from the under side even though glued to a strip of mica. Strips of blue paper were pasted on the mica to reduce the glare and afford a possible place to enter a small amount of data or a number. Mica has given way to some use of cardboard in most instances. Heat has a lesser effect upon the paper and curling is not experienced. These points may be cut from cardboard with a pair of scissors by ruling a series of triangles 3mm. wide and 7 mm. high, or they may be made with a specially constructed punch. If a good punch is at hand nice points can be made, but if the punch fits loosely the points may have an objectionable fringe on one side. Glue, shellac, or some specially prepared adhesive may be used to mount the specimens on these points. Here again opinions vary and about the only point of agreement is that the specimens should be mounted so that when the point extends to the left of the pin the head of the specimen should extend forward. Some workers prefer to use very fine points, others more blunt points in their efforts to leave the greater part of the under side of the thorax exposed. Some prefer to mount two specimens on one point; one on its ventral and the other on its dorsal side. Dr. Frison in the Illinois State Natural History Survey uses double points to mount elongated forms, such as May flies. Gelatin capsules have been used but they become hard and brittle after a few years and specimens cannot be studied without removal from them. The pin is

usually passed through the capsule and cap after the latter has been slipped on.

Materials used for supporting structures used in double mounting have been varied indeed. Small cubes of corks are most common, while pith, soft wood, certain fungi and other materials are encountered.

For larger specimens the methods of pinning are not so diverse. Earlier collections were pinned low on the pin but this practice is no longer followed. Specimens are placed near the top of the pins. The exact position is still a controversial matter, but the main considerations are that the insects should be placed at such a height on the pin that they will not be injured when the top of the pin is grasped by the fingers in handling the specimen and that all specimens are all arranged at the same height on the pins. In general, an insect should be pinned through the thorax along the median line, altho Coleoptera provide a marked exception to this rule. They should be pinned thru the anterior portion of the right elytron. Hemiptera should be pinned through the center of the scutellum. There is a general tendency to slant the pin slightly forward when inserting it in the body so as to include two segments giving greater strength to the body and giving a slight tilt to the specimen. Opinion differs in this regard, too, and the essential thing is that one should be consistent in which ever method he follows. Older workers carefully spread the legs of specimens, but this practice is not so strongly adhered to now. The tendency is to allow the legs to double up under the bodies to conserve space and prevent damage in handling. Care should be taken, however, to see that structural characteristics of the under side are not obscured by the legs. In groups where the mouth parts serve as characters for classification, it is well to see that they are visible while they are still movable. This often saves time in relaxing later.

It is advisable to spread the wings of certain insects in order to provide for visibility of certain characters for use in identification and to add beauty and uniformity to the pinned collection. Spreading boards have been devised to save time in this operation and variously modified types are available at moderate cost at supply houses. Rather simple boards may be constructed and directions for making such boards may be found in Banks "Collecting and Preserving Insects," U. S. National Museum, Bull. 67, page 58. Glass plates may serve as spreading boards. Place two

glass plates side by side leaving a crack just wide enough to admit the body of the insect. Place the insect in position and after spreading the wings of each side to the desired position use a smaller glass plate to hold them in place. This method is especially suitable for spreading Lepidoptera destined for the riker mount where there is to be no pin in the thorax.

The wings of both sides of Lepidoptera should be spread and only those of the right side of Orthoptera. The other pair should remain as indications of the position taken at rest.

The apparatus used in pinning insects is not necessarily elaborate. Two pairs of forceps, one for picking up small insects and the other for setting pins; a pair of scissors; a spreading board; some strips of paper; some points; and a supply of variously sized insect pins are all that are absolutely necessary. A pinning block is quite handy for getting specimens at the same level on the pin and in placing labels at varying heights. In absence of a pinning block a small cardboard box is a satisfactory substitute and many workers, especially in the field, use the lapels of their coats when pushing the pin through the specimens.

There are many insects or insect forms for which pinning is a very unsatisfactory method of mounting. Some of these can be mounted in stoppered glass containers in some preserving fluid or in some instances as dry mounts. Materials used as stoppers include cork, rubber and glass. Cotton may be used in temporary mounts, where the specimens are not completely dry. Cork is satisfactory as a stopper provided a good grade is used. It is easily available and is fairly economical in price. Evaporation is a great factor when cork is used and periodic examinations should be made of all containers to replenish the evaporated preservative. Rubber stoppers reduce the evaporation to a marked degree although they are quite expensive and when in contact with alcohol, sulfur crystals often form and become lodged on the specimens. This can be prevented to a marked degree by soaking the stopper in hot water for some time prior to use. Rubber stoppers especially should have a small insect pin inserted along their sides to allow for the escape of air or liquid displaced by them. When the stopper has been inserted to the desired position the pin should be withdrawn. Stoppers so inserted are less likely to "pop-out" with temperature changes than those inserted in other ways.

Vials are the most common perhaps of stoppered containers and are chiefly of three kinds: homopathic, shell and ring vials.

They are obtainable in various sizes and are useful in many ways in preservation. For specimens not in constant use the vials may be corked and dipped into paraffine wax as an aid in the prevention of evaporation. If properly done this method is quite satisfactory.

Bottles are used similarly to vials, but have the advantages of more sturdy construction and greater size. Both bottles and vials are useful in collecting and museum display work.

Mason and museum jars are other forms of stoppered containers. The Mason jars have screw or spring clamp lids which fit down tightly over rubber rings to reduce evaporation to a minimum. Even then it is necessary to make periodic examinations of materials mounted in them to replenish preservative material where necessary. They are of greatest value to the entomologist for storing his duplicate material in bulk and for storing small vials to reduce evaporation. Biological supply houses find them useful and various sizes to suit particular needs are obtainable. Museum jars are useful in temporary display of specimens, but evaporation is too great from them. There is a small hole in the lid of each that is supposed to take care of pressure changes to prevent breakage and this allows too much evaporation. Various waxes have been used to seal them and murrayite, an adhesive insoluble in alcohol or water, promises great usefulness in preventing this evaporation. Ordinary sealing wax is soluble in alcohol after a time.

Tubes and vials are quite similar in their adaptation. Tubes are usually longer and the ends are rounded which makes it hard to stand them anywhere for display. Various attempts have been made to include alcoholic tubes with the regular pinned collection and special blocks have been devised with spring catches for holding these tubes. Comstock devised a square bent neck vial that was used somewhat. In any case where stoppers are used a great deal of grief comes from evaporation and the stoppers sometimes come out allowing the liquid to run over the box. The Illinois State Natural History Survey made up a collection of insects a number of years ago and sent them to high schools. These included some alcoholic mounts that were sealed in a flame by Dr. R. D. Glasgow, then of the University of Illinois. These were satisfactory for their purpose but it is doubtful whether it is advisable to include alcoholic and pinned specimens in the same box.

Elaborate apparatus is not necessary for making stoppered glass mounts. Forceps, pins, needles, stoppers, preservatives and labels are about all that are necessary.

Hermetically sealed tubes make very satisfactory and attractive insect mounts. The specimens are placed inside, secured in some way to prevent damage in handling, and labeled and sealed. A similar method was in use in 1887 by Dr. H. Dewitz in Germany. He mounted the larvae of Microlepidoptera in very small tubes which he made from long tubing and sealed by use of an alcohol spirit lamp. The finished tubes were mounted in a hole in a cork and pinned with the related specimens. There was scattered use of sealed tubes from then until about 1910 when Dr. R. D. Glasgow began to develop the method at the University of Illinois. Some material was sealed for the State Natural History Survey and soon attention was directed to the department collection for class use. This work has progressed and it has been the privilege of the author to work with Dr. Glasgow and to carry on the work after he accepted a position in New York. In personal correspondence with Dr. R. K. Nabours at Kansas State College the information was received that grouse-locusts sealed in this manner about 15 years ago are in good condition. The method in use at Kansas is not so elaborate as at Illinois according to Dr. Nabours.

This method admits a wide range of specimens and a variety of ways of mounting each. Both dry and liquid preservations may be used and one can use his own initiative in deciding upon the method to follow. One somewhat skilled in glass working can modify the general methods considerably as glass supports are very neat and serviceable. Certain insects because of large size or other reasons need not be held by special supports. In many cases where no particular structural character is to be sought in later study there is no particular value in the extra labor involved of putting the specimens on supports. Larvae for the most part should be supported in some way while many adult specimens make much more serviceable mounts if supported in a certain position. Pins are used to a limited extent in hermetically sealed tubes. They may be bent before or after the insect has been placed upon them and then stuck into a cork triangle or fused to a glass rod in such a manner that the specimens assume the desired position in the tube. The cork triangle method is inferior to that of the glass rod. The pins become loosened in the cork and the triangle sometimes

slips leaving the specimen in an unsightly position in the tube. In fact, the use of pins in the glass tube method is not so satisfactory. There is too much tendency for specimens to slip off or turn and assume positions which make their study impossible. If pins are used they should be fused to a glass rod around which cotton has been wrapped to form a neat plug (see p. 311). The pin and rod should both be heated and the pin thrust into the end of the rod while both are still hot. If the enamel has been burned off, the pin will fuse with the glass and a permanent mount will result. This cannot slip sidewise as the cotton plug, properly rolled, prevents that and a shock great enough to break the glass rod would probably break the tube also. This method furnishes a means of transferring valuable pinned specimens to tubes where the damage from pests is impossible.

Glass rods alone form a very important group of mounting structures. Small solid rods ranging from about 5 mm. to 8 or 10 mm. in size are most convenient. These rods should be cut into convenient lengths and placed in a box for use later. For some forms a straight rod is used. This is made by heating a short section of the rod in the flame of a blast lamp (see p. 313) and drawing out to the desired thickness. The rod may be pointed for insertion into the specimen by placing in the flame at the point where it is desired to cut it off and drawing out rapidly. Bent rods are made in much the same way except that there is a right angle bend made in the glass rod before pointing it. Rods with double points are useful in supporting long and weak specimens, whose form makes it impractical to use the straight rod. These rods are made by welding a piece of glass rod to the bend made for the first lateral point. A second point is now made as was the first one.

Mica plates, opaque glass, cardboard and other materials have been used in mounting various insects. They are cut to fit the inside of the tube and the specimen is glued to them. Life cycles have been mounted in this same manner.

When liquid preservatives are used it is necessary to leave a small amount of air space to prevent changes in temperature from breaking the tube. This air bubble slips past the cotton plus that holds the label and specimen in place and interferes with the visibility of the specimen. In most instances it is impossible to remove the bubble through the plug and provision for its removal must be made while the tube is in process. This is best accom-

plished by a valve around which the cotton is wrapped to hold it in position. Glass rod supports may be welded to the large end of the valve when necessary. These valves are made by taking a piece of glass tubing about one-half the diameter of the tube to be sealed and cutting it into pieces about two inches in length. With the blast lamp adjusted to a fine and intense flame, rotate the tube in it at its center until it begins to melt. Remove from the flame and draw out rapidly yet carefully enough that the center of the drawn portion corresponds with the center of the tube until the drawn portion is slightly less than half the diameter of the original tube. With a knife or sharp file scratch the drawn portion at its center and break. Thus two valves have been made with one operation.

The cotton plug mentioned above serves a double purpose. It holds the label and the specimen support. It is necessary, therefore, that it be so constructed that it will not have side motion. The plug is made from a strip of absorbent cotton. It may be medicated or fumigated but one or the other should be used to prevent the introduction of insect enemies that may harbor in untreated cotton. A strip of convenient length about 3/16 of an inch thick and slightly narrower than the label to be used should be torn from the roll. With the fingers of both hands gently pull the cotton sidewise being careful not to tear it completely. When this is done roll the edges in toward the center until the width of the resulting strip is just that of the label. If no support or valve is to be used, roll a loose yet firm plug that will fit snugly inside the tube. If a valve or support is to be used, place a drop of glue on the support and roll the cotton around it until it fits snugly inside the tube after the label has been wrapped around it. If these operations have been carefully done there will be no loose edges of cotton sticking out from under the label and the plug will strike the glass with even pressure at each end thus preventing any side motion from shocks in handling.

For special mounting of some larvae and other elongate forms a constricted end tube is sometimes used. This mount is made by rotating a tube in a wide hot flame and gradually and carefully pulling out until the desired size is reached. This must be carefully done to insure the center being in line with the center of the larger tube and to maintain an even thickness in the drawn portion. Scratch the drawn portion at a desired point and break. Then thrust into the flame again and re-blow a new end on the

constricted part. After cooling place the specimen in position, add preservative and label and seal.

The difficulty is often experienced that insects fall from their supports after varying lengths of time. This may be prevented in a number of ways. With dry mounts ordinary glue is usually satisfactory. The muscles of insects contract and set after death and advantage may be taken of this fact to insert the supports before they have set. Thus the tissues set around the support and there is slight chance for their falling off. Slight heating at the point of entrance of the support often helps in this same way. For liquid mounts murrayite or some other adhesive insoluble in the preservative used should be employed. The author has had satisfactory results with celloidin as an agent for holding the eggs of aphids on bark. The eggs are coated with celloidin and the piece of bark dropped in 70 per cent alcohol before the celloidin has had a chance to harden. The visibility is not impaired in any degree and the eggs remain on the bark in life-like position. This method could be adapted to other things with equally good results.

There are no hard and fast rules governing the size and intensity of flame to be used in sealing tubes. It should be wide and intense enough that the glass can be drawn out without leaving a square shoulder next to the plug. However, it should not be heated in such a manner that the shoulder breaks off when cool. The tube should be held in such a position that it can be freely rotated in the flame until it almost closes off with molten glass. Then remove from the flame and draw out in such a manner that the drawn portion will remain at the center and its diameter will be about 2 mm. When cool break and while rotating in a vertical position close off the opening with a needle point flame. This method is the same for the liquid mount except that the preservative is usually inflammable and a certain amount of extra care is necessary to prevent breakage. The last process is usually the hardest. The preservative is close to the end of the tube and being very volatile generates gas very rapidly. This pressure causes bubbles in the molten glass and quite often causes the tube to break with explosive violence. A few trials will show the operator when to remove the tip from the flame to prevent this bubble burst. It may be said in general that it should be removed just as the molten glass begins to close over the opening.

The apparatus necessary for making hermetically sealed tubes is rather complicated. In the first place three kinds of glass are necessary. Soft glass tubes with regular ends sold under the names of ligature or culture tubes should be on hand in various sizes to accomodate any size of insect specimen. Various lengths of soft glass tubing for making valves should be on hand and a supply of variously sized solid glass rods should be available.

The blast lamp is quite important. On its ease and fineness of adjustment depend some of the finer points of the actual sealing operation. It should be capable of adjustment from a fine needle flame to a wide intense flame for heating large tubing. The Massachusetts Institute of Technology pattern has three interchangeable burner tips with which the range of flame is very satisfactory. A supply of oxygen should be available for use when sealing very large tubes and should be attached to the compressed air line.

A glass cutter is quite essential to economy of time in producing valves and other glass apparatus. A satisfactory electric cutter is in use in the laboratory at the University of Illinois. The cutting unit is a piece of No. 22 gauge chromel wire attached to the regular 110 watt circuit. There is a resistance unit composed of a salt solution (10 gr. salt to 1000 cc. water) through which the current must pass. A ring is made in the chromel wire to fit snugly around the tube at a point where a scratch has been made by a file or knife. When the current is turned on by a convenient switch held in the hand the wire becomes hot quite rapidly and the glass breaks squarely at the scratch. If the heating occurs rapidly enough this apparatus gives satisfactory results with glass of ordinary thickness. Too slow heating melts the glass and does not break it squarely.

For short tubes provision must be made to prevent burning of the hands while sealing. Cork is a good heat insulator and is quite satisfactory. A triangular trench is made in one side of a cork to allow for the escape of gases generated by heating and the tube may be turned by turning the cork. The author has used a double cork which is fastened together with staples made from insect pins. This affords a round surface to hold while turning and prevents occasional burning when the gas ignites at the end of the tube.

Push rods are necessary to insert the specimen into the tube to its proper position. Glass tubing just small enough to enter

the tube being sealed is best since equal pressure is exerted on all points of the circumference of the label and it remains straight.

Various needles and forceps are necessary. A needle with a screw end is quite handy for twisting into the cotton and withdrawing the plug if it becomes necessary to do so.

The atomizer bulb is very useful in filling tubes with preservative after they have been drawn out. The bulb is filled with liquid and appressed to the open end of the tube. If compressed a few times the tube can be easily filled. The contents of tubes can be quickly removed by inverting them and compressing an empty bulb against the open end. Air is forced into the tube which replaces the liquid.

Stuffing has also been used as a means of mounting and preserving specimens, but is no longer used to any extent. Bruner has given a full account of stuffing Orthoptera for museum cases.

Inflation of larvae is another form of mounting that is no longer in general usage. The operation is complicated and the specimens are very easily broken after they are prepared. They can be secured from supply houses almost as cheaply as they can be made unless one is especially equipped for inflating them.

Riker mounts are useful in a number of ways. They are of pasteboard construction fitted with a glass cover and filled with absorbent cotton. The bottom should be covered with naphthalene flakes and the cotton replaced. The specimens should then be put in and the cover placed over them. Binding with binding tape is essential to keeping out museum pests. The General Biological Supply House at Chicago has developed a metal box of similar size called the Turtox Metal Mount. It is more durable and binding tape sticks to it more satisfactorily than to the pebble grain finish on most Riker mounts.

V. METHODS OF LABELING THE INSECT COLLECTION.

The methods used in labeling the insect collection are varied. The labeling depends upon the kind of collection and the personal whims of the workers. Some methods used by certain workers are hardly understandable to other workers. One of the principal methods of labeling collections has been to assign each specimen a serial number and enter data under this number in a notebook. Certain amounts of data are necessary for the classification of some insects to species, and collections so labeled are almost worthless if the reference book is lost. The opposite extreme is found among

other workers, especially in other lands, where all the data known about one specimen are attached to the pin below it. The data are written upon sheets of very thin paper and the sheets folded to a compact square before the pin is inserted. Most workers now agree that certain amounts of data should accompany each specimen and that the data should be in such a position on the pin as to be easily read without removing the specimen from its box. It is conceivable that detailed data cannot be given if this rule is to be followed; so a combination of the above types of labeling is growing in favor. The locality, date, and sex labels with the name of the collector should accompany all specimens. A serial number is placed below the collector label, and complete known data are entered under this number in a card index or notebook. Thus the specimen can be identified positively, even though the record indicated by the serial number is lost. As soon as determination to species has been made it should appear as the lowest label and if determination was made by a specialist or if the specimen has any special significance such as having been used as a type, this should be recorded on the name label or on a separate one above the name label.

Opinion is varied as to the amount of information that should be included on the name label. In most cases perhaps the best procedure is to include only the genus, species, and author of the species. In cases where varieties are recognized these names should also appear.

Specimens for class room purposes may well be designated by serial numbers. These numbers can be recorded in a card index along with the name and any known data concerning the specimen. The numbers are an advantage when giving identification tests.

Specimens preserved in liquid should have the labels inside the containers. They may be placed outside, but they often come off and become illegible from some cause or other. Ordinary inks wash off in preservatives; so India ink must be used. Lead pencil may be used for semi-permanent labeling. The sealed tube method affords an excellent means of placing the labels on the inside in such a position that they are readily visible and at the same time do not obscure the specimens in any way.

Labels may be made by a printer who is equipped with very small type, or they may be secured at supply houses at a nominal cost. If one can print nicely he can print his own labels. How-

ever procured, they should be on a good grade of paper and should be printed with permanent ink. This is especially true of labels intended for use in liquid mounts. Beamer ('16) has published an article in which he describes a photographic method for making labels from a typewritten copy. The original is made up with desired spacings and the reductions in photography made as desired. The individual labels are cut from the resulting print.

Each specimen of insects in a collection should have a complete set of labels. Workers modify this rule to suit themselves by placing complete labels on only one specimen in a group of the same species. The name label is the only one that is usually omitted and this for the most part in cabinet collections. Duplicates may very well be labeled in this manner to save time, but there is much to be gained in having each specimen in a collection bear complete labels. There is a great need for uniformity in labeling collections. Workers who expect to trade material should at least be consistent in labeling the material collected by themselves.

VI. PRESERVATIVES

Alcohol is probably the most generally used preservative. It is used alone in strengths varying from 50 to 95 per cent or in combination with other preservatives. Specimens placed in it should be started in lower grades and "stepped up" to the percentage in which final preservation is to be made. It is not especially good for histological work, after preservation for longer than two or three days.

Formaldehyde is recognized as a better preservative for general use than alcohol, but it has a strong odor and affects the eyes and hands if specimens preserved in it are to be studied later. For museum displays and other mounts not intended for study it has a definite use. It is used in strengths varying from 4 per cent. to 38 per cent. Formalin is the trade name for 38 per cent formaldehyde in water and this name is sometimes used instead of formaldehyde 38 per cent. Tillyard uses 4 per cent formalin as a killing and fixing agent and 38 per cent is often used in injection work.

Glycerine has a softening affect upon materials preserved in it and does not penetrate well enough to preserve internal structures. Thus specimens so preserved often become discolored from deterioration. When mixed with alcohol it makes a good preservative, and prevents dessication of specimens if the alcohol evapor-

ates off. Lee has recommended a preserving fluid made from equal parts of 95 per cent alcohol, glycerine and distilled water. He says that specimens preserved in this way may be used for histological purposes or may be used for gross dissection. The author has used material thus preserved for gross dissection and the results are pleasing.

Tothill has used chloral hydrate as a preservative. He killed his specimens by pouring hot water over them and allowing them to drain over a screen or blotting pad for five minutes after which he placed them in a container and poured three times their volume of chloral hydrate (5% aqueous solution) over them and closed the container. After a week he changed this solution to a fresh one of the same strength and left for permanent preservation. He says that he has secured fine results with histological studies made from specimens preserved in this manner and that after four years the specimens were in good condition.

Many other substances have been used as preservatives but few of them are practical now and will not be mentioned here.

VII. Preservation of Color in Insects.

Tower ('03) in an important contribution to the knowledge of insect coloration says that color patterns in insects are of two types: one as old as the insects themselves is composed of cuticular and hypodermal colors in segmentally arranged spots and stripes corresponding somewhat to the position of vital organs; the other, phylogenetically younger, is composed of scales and hairs superimposed upon and partially obscuring the older type. The second type is of purely ornamental value and is quite diverse in arrangement and coloration, while the first type is of more importance. The colors of this type have been designated as "colors pertinent to the species."

Colors in general are of three classes: chemical, physical and and chemico-physical. The chemical colors are divided into three groups: cuticular, hypodermal and sub-hypodermal. Those of the cuticula are black, brown, dark brown, and straw yellows and are located in the primary cuticula. They are permanent, being soluble only in strong mineral acids. The hypodermal colors are subdivided into two groups depending upon their derivation and location. (1) Those located in the hypodermal cells as granules and caused by lipochromes include the chrome yellows, red, vermillion, scarlet, and blue. They are insoluble in water and

ordinary preservatives, but are soluble in ether or other fat solvents. (2) Those located between the hypodermal cells are derived from pigments. They are not permanent. They fade at death or exposure and are soluble in water and ordinary preservatives. They include the colors green, yellow, and white and are largely derived from chlorophyll and xyanthrophyll. The sub-hypodermal colors are green, yellow, and white and are located in the haemolymph or fat body. They, too, are derived pigments and are soluble in water and ordinary preservatives. They fade at death or upon exposure.

The physical colors include white and the irridescent and metallic colors. White unites with other colors to form a sub-group of opalescent colors. Pigment is usually present. The physical colors are produced by surface modifications which reflect, refract or diffract light. They are closely associated with the colors of the next group.

The chemico-physical colors occur widely and are the most brilliant and varied of those produced in insects. They are divided into four groups: (1) reflection pigmental colors, composed of blacks, browns, yellows, and reds and caused by reflection from a polished lamella over a layer of pigment; (2) refraction pigmental colors which include almost all metallic colors, caused by a polished refractive lamella overlying a layer of pigments; (3) defraction pigmental colors in which colors of the above three groups combine to form certain color effects. This group is confined almost exclusively to insects bearing scales.

From the above outline we find that the colors usually retained in preserved specimens are those whose escape is impossible because of insolubility. Few or none of the green, yellow, or white colors are preserved and many of the chemico-physical ones are lost because the pigments essential to their production are lost in preserving methods now in use. In many cases the hypodermal reds, yellows, and blues are dissolved out through the use of ether. Preservation of insects has been done too much by blind chance methods. Insects were placed in preservatives and the colors either stayed or were lost without anyone knowing particularly why. The above work points the way in the opinion of the author to the retention of the most elusive colors. They may be attacked by a chemical method and rendered insoluble in the ordinary preservatives, or an entirely new preservative must be produced.

VIII. METHODS OF HOUSING AND DISPLAY OF COLLECTIONS

Various groups of insects require different methods of housing and displaying as we can readily see from the various methods used in mounting. The pinned collection is ordinarily placed in boxes of various construction and stored in many ways. Individual collections are usually pinned in Schmidt boxes (see paragraph below) and must be opened for reference while larger collections are often housed in boxes with glass tops and placed in large cabinets or arranged in frames for display.

The Schmidt box is a very serviceable one. The top and bottom are two thin pieces cross-grained to prevent warping. The top fits down tightly over a raised ledge of the lower part, making the entrance of pests quite hard. They are made of seasoned pine, white-wood or linden and are 8½″ x 12″ in size, altho a size 12″ x 15″ is also for sale. The bottom is lined with pressed or patented cork. Sheet cork is much better, but more expensive and a later recommendation is the use of Celotex, a composition fibre board used in building construction. It does not rust pins and does not bulge as is often the case with cork.

The U. S. National Museum uses a drawer 2½″ x 18″ x 18″ which fits into a metal case. The box has an inside partition enclosing a space about ¼″ wide all around which is filled with naphthaline to form a poison chamber. The sides and top frames are made of well-seasoned wood and the bottom is of three-ply cross-grained veneer covered with patent cork and lined with white paper. The cover is of glass, fitting into a frame ¾″ wide with a ¼″ tongue which fits into a groove between the inner and outer boxes. A groove is made upon the sides of the boxes to correspond with sliders in the cabinets.

"Professor Comstock of Cornell University has adopted a case with a top and bottom of glass. Its outside dimensions are 16″ x 19″ x 2″ and the covers are both dovetailed and mitered. The top and bottom are alike except that the former is not quite so deep and is grooved to fit over the tongue of the latter. The bottom is covered by a series of wooden blocks each about ⅓″ thick. Twelve of these unit blocks completely fills the box. There are various sizes adapted to fill varying needs, the idea being to put all of one species on a block, thus avoiding the necessity for repinning in case of reorganization of the case." Felt ('99). A decided improvement in this type of box is the substitution of a good grade of sheet cork or Celotex for the board blocks.

The above described boxes are all rather expensive and there is now advertised a box called the Hood Insect Box. It inside dimensions are 8¾″ x 12¾″ x 2¾″ and the frame and cover is made from three-ply veneer with interlocked corners which enables the cover to fit tightly upon the box. The pinning bottom is composed of sugar cane pulp instead of cork and the box is lined throughout with white glazed paper. The outside is covered with water grained heavy black paper making a very neat box. One of the greatest claims of its advertisers is that it is pest proof.

Other boxes have been used by various workers to satisfy particular needs but these are not generally used and will not be discussed here.

Pasteboard boxes are in general use by amateurs because of their cheapness. Other workers sometimes use them to house duplicates. They are variously sized and constructed and are for the most part quite frail. Pests enter them very readily.

Some workers use a loose-leaf method for mounting insects and there are various modifications of the Essig ('26) model. The insects are mounted in cavities cut in corrugated paper and covered with strips of mica. When complete the whole mount is covered with white paper in which holes have been cut to correspond with the mica strips. The data for each specimen should be placed near it on the white paper. This is not a generally used method and does not appear to the author as a very practical one.

Lepidoptera are often mounted in private collections in book form. Some of these are quite elaborate and allow for expansion as new species are collected. A detailed description of one of these occurs in Entomological News 37:282, 1926.

IX. Enemies of the Insect Collection

Collections are often set aside for reference or stored away after complete and careful labeling and arrangement without taking proper precautions to prevent attacks by various enemies. There may be local factors working to destroy collections, but chief among the recognized enemies are: (1) museum beetles; (2) book lice; (3) confused flour beetles and other small Coleoptera; (4) greasing; (5) verdigris; (6) mould; and (7) mice.

Museum beetles, of which six species have been reported on insect collections, are perhaps the most serious enemies of the collection. They may occur in collections housed under the very best of conditions and they work rapidly when once established.

Some workers contend that the eggs are laid by the adult beetles upon the boxes and that the larvae crawl through cracks in the boxes while others think that the eggs must be laid directly upon the specimens. Collections that have been closed for years often become infested, but it is not clearly understood how these minute larvae crawl up the enameled surface of the pins in order to reach their food. The larva is the most dangerous stage in the life cycle of these pests, and under favorable conditions the cycle is continuous. The larva feeds within the body of its dried host until food material gives out, or until pupation and emergence. The excrement is pushed outside the host through the entrance hole and it is by this fine brownish dust on the floor of the boxes that the presence of these pests can be detected. Infected specimens, or preferably whole boxes bearing infested specimens, should be removed to a fumigating room and cared for (see page 323).

Museum beetles belong to the order Coleoptera. Three genera of the family Dermestidae are represented. *Anthrenus museorum* L. is an important species and prior to 1890 was found only in collections recently imported from Europe. *Anthrenus verbasci* L. is a common American species and is not so injurious as *Anthrenus museorum* L. *Trogoderma tarsalis* Melsch and *Trogoderma ornata* Say replace the species of *Anthrenus* in the southwest. *Attagenus pellio* L. has been known to inhabit insect collections in the eastern part of the United States, but its chief damage is to furniture.

Book lice are small soft-bodied insects of the family Atropidae of the order Corrodentia. They may be wingless or may possess vestigial wings. They are usually found in collections that have been stored in warm moist places often accompanying mould which will be discussed in a following paragraph. Three species are recognized by Tillyard ('26) as affecting insect collections: *Atropos pulsatoria* Mull., *Lepinotus inquilinus* Heyd. and *Troctes divinatorius* L. They crawl about over the dead insects and eat spines, hairs and lesser chitinized cuticula which with smaller insects may prove to be quite disastrous. They succumb easily to fumigation and may also be killed by dry heating.

The confused flour beetle and other small Coleoptera are rarely serious enemies of the insect collection as dried insects are somewhat removed from their list of food materials. *Ptinus fur* L. is

reported by Riley ('92) as a pest of entomological specimens, but he reports it as being more common in Europe. Blatchley in the "Coleoptera of Indiana" reports this species as being widely distributed by commerce and often injurious to museum specimens. It is more especially fond of furs. *Tribolium confusum* Duval and *Tribolium ferrugineum* Fab. are reported by Riley and Blatchley as doing more or less damage to insect specimens, but these are more closely associated with stored grain products.

Greasing often damages particular groups of insects, chiefly Lepidoptera, Coleoptera, some Hymenoptera and some larger Diptera. It is caused by the melting of fat globules released by deterioration or rupture of the cell walls which retain them. The affected specimens present a characteristic oily appearance and soon become dirty because of the adherence of pieces of lint and particles of dirt from the atmosphere. The colors, scales, and pubescence are likely to become damaged, thus reducing the value of the specimen in the collection. Insects affected with greasing need not be discarded if discovered in time. Muschamp ('12) has devised a means of removing greasing with toluol which he characterizes in his article as "a new solvent." He takes three containers with covers and places toluol in each. Specimens are started in the first where they remain for 24 hours. Most of the grease is loosened in this bath and by two successive baths of 24 hours each he removes all of it. After drying, the specimens are as good as new. A decided advantage for his method is that toluol does not relax the insects and it is not necessary to reset them after these baths.

As a preventive for greasing, St. John ('15) recommends the setting of Lepidoptera over a sponge of cotton wool saturated with formalin. His contention is that the fumes of the formaldehyde enter the bodies and prevent greasing. Gilles ('15) substantiates his views in this process and explains that formaldehyde unites with the chitin and cell walls and prevents their deterioration when in contact with moisture. This, he says, prevents the release of fat globules and thus prevents greasing. His methods differ slightly from those of St. John and he advises that formaldehyde is injurious to the colors of the emeralds, but that other colors remain unchanged. Later workers have shown rather conclusively that only those colors within the chitin will withstand the action of formaldehyde for very long.

Verdigris has been a more serious enemy of the insect collection than it is today. It is caused by the action of certain body juices and acids upon the pins and appears as a bluish corrosion at the dorsal and ventral union of the body and the pin. It has no particular effect upon the insects but the pin may become brittle or eaten away and break. The older white pins were more susceptible to verdigris than the common japanned or enameled ones in use today. The fact that greasing and verdigris are often found on the same specimens has led to the belief that greasing was a cause of verdigris. Greasing is caused by fat globules melting and running over the body while verdigris is the result of the corrosive action of certain body juices and acids upon the pins.

Mould is a serious enemy of the collection in some instances. If stored in a warm moist place mould is likely to occur and collections so infected are very hard to clean so as to be valuable at all. There are probably several fungi causing this condition. A species of Aspergillus has been found in a collection at Urbana, and there are several others common in the air that would thrive under similar circumstances. Ordinary repellents are said to prevent mould as well as insect enemies.

Mice, while not usually recognized as an enemy of the collection, may cause serious damage if specimens are left exposed to dry, or if the boxes are accidentally left open.

X. Fumigation of the Insect Collection

In the control of enemies of insect collections it often becomes necessary to resort to fumigation. Some workers take the stand that fumigation of the collection is one of the necessary parts of its care and that it should be fumigated periodically. In some areas where the evaporation rate of the commonly used repellents is high, this is probably necessary, since the usefulness of the repellent may be spent (before the owner is aware of the fact) and certain enemies may have entered by the time the owner becomes aware that his supply of repellent needs replenishing. Of several well-known and recognized fumigants for this purpose, carbon bisulfide is the most widely recommended. There are various methods that may be used to fumigate collections with carbon bisulfide and where one has a large collection it is advisable to build a tight container or room of known dimensions where several or all the boxes of the collection may be placed for fumigation. The boxes should be opened and placed in such position that the

gas can enter them readily. As soon as everything is arranged, saturate several cloths with the carbon bisulfide and place them in the chamber, preferably near the top as the gas is heavier than air and will press downward. It has been found that if applied at the rate of one pound per 100 cubic feet for 24-36 hours very satisfactory results are obtained from carbon bisulfide.

Some workers prefer to fumigate boxes individually. This is accomplished in two ways and has at least two distinct disadvantages. The first way is to pour into the box a quantity of carbon bisulfide. This method embodies both disadvantages: the liquid stains the floor of the boxes and causes the pins to rust. The second method is to pour the fumigant into a watch glass in a corner of the box. This, if carefully done, removes the staining disadvantage and to some extent the rusting, but the excess moisture occasioned by the addition of the fumigant does not escape readily as the boxes must be tightly closed for 24-36 hours. The individual box method has advantage in the saving of time and the ease with which it may be done, but the preferable way is the one mentioned above. Still others prefer to remove infested specimens to separate boxes for fumigation. This is a good practice if one can be certain that he is removing all the infested specimens. This process may prove to be long drawn out because of the several stages in development of the pest.

Carbon bisulfide has other disadvantages as a fumigant. It is highly inflammable and explosive and must not be used near a flame. Even sparks from switches of electric systems may cause explosions. It is poisonous but its disagreeable odor probably prevents one from becoming seriously injured by its use. This odor is quite disagreeable and forms a third objection to its use as a fumigant. Since the boxes must remain closed, the odor remains for a very long time and is, to some at least, a serious disadvantage especially when the boxes must be opened occasionally or studied. Despite these disadvantages carbon bisulfide is probably the most widely used fumigant for insect collections.

If one is properly equipped for its use hydrocyanic acid gas makes a very good fumigant. This requires very careful treatment as it is a most deadly gas. It was formerly generated by the action of sulfuric acid and water upon sodium cyanide in an earthenware jar, but there has recently appeared a calcium cyanide dust which, when exposed to the atmosphere, liberates hydrocyanic acid gas. There are two kinds of this dust according to Metcalf

and Flint ('28). The 88 per cent dust should be applied at the rate of ¾ pound to each 1,000 cubic feet of space and the amounts for the 48 per cent dust should be about doubled for the same space. This dust should be applied by spreading it about ⅛ inch thick upon newspapers. The fumigation chamber must be kept tightly closed and provision should be made beforehand for opening and ventilating the chamber after the process is complete.

Paradichlorobenzene is also used as a fumigant. It should be resorted to only when danger of fire is great. It has only a fair killing power, and is more expensive than other materials. It may be applied in porous cloth sacks or in tubes covered at each end with fine gauze.

Carbon tetrachloride is also used, and as with paradichlorbenzene, should only be used in cases where the fire hazard is very great. Its killing power is very weak and it is very volatile which makes it a poor fumigant and bars it entirely from use as a repellent.

Older entomologists have used a variety of materials as fumigants largely, perhaps, because of their disagreeable odors. Most of these we now know have only repellent values. Chief among these are found such substances as camphor balsam, oil of turpentine, and forms of creosote.

XI. Safeguarding the Insect Collection

From the discussion in Section IX we may draw some conclusions here as to the methods for safeguarding collections from these various enemies. Perhaps the first thing to be done after one has made certain that all pins are set securely and that specimens loose on pins are securely braced is to see that the boxes are stored in a dry cool place. This will prevent the entrance of mould and book lice and will lessen the amount of greasing to an appreciable degree. It will also remove the probability of warping of boxes as is often experienced in more moist localities. The boxes should be of as tight construction as possible both with regard to fitting of lid and corner construction. Dermestid larvae can make their way through very minute cracks. A tropical method of safeguarding against pests is to stretch a rubber band around the box at the point of union of the box and lid. This prevents the entrance of almost all minute enemies.

Even though all these precautions are taken, a liberal use of some repellent is advisable. Naphthalene cones are available from

supply houses or ordinary moth balls mounted upon pins may be stuck into the boxes as desired. If the head of an ordinary insect pin is heated to red heat in a flame and thrust into a moth ball the naphthalene that is melted by the heat will adhere to the pin upon cooling and the ball may be set anywhere desired. Para-dichlorobenzene is often used, but it is too volatile and expensive. Creosote is not so good because of damage of soiling the box or specimens if it should be spilled. Camphor balsam or carbolic acid is an aid in prevention of mould. Mercury pellets are said to pick up particles of dirt as they roll about and are also said to act as repellents.

If one observes all these precautions his collection may escape infestation, but periodic examinations should be made to detect any signs of damage. It is a case where a small amount of work may mean the saving of a valuable collection. These repellents are good and their value has been proven, but too much emphasis cannot be placed upon the fact that they sometimes fail. Their strength may deteriorate just to the extent of allowing a pest to enter, yet the odor may seem strong enough to be effective.

If specimens are received from other collections in exchange these should be subjected to fumigation and a period of probation before being set in the collection.

BIBLIOGRAPHY

BANKS, N.
1909. Directions for Collecting and Preparing Insects. U. S. National Museum Bulletin 67: 1-135.

BEAMER, R. H.
1916. An Easy Method of Making Insect Labels. Entomological News, 27: 419.

BRUNNER, L.
Directions for Collecting and Preserving Insects of Orthoptera for the Cabinet. Univ. of Neb., Dept. of Agr., Bull. 42: 1-8.

CHAMPLAIN, A. B.
1928. A Method for Shipping Insect Collections. Bulletin Brooklyn Entomological Society 23: 253-254.

COCKERELL, T. D. A.
1897. Directions for Collecting and Preserving Scale Insects. U. S. National Museum Bull. 39, part L; 3-7.

CRESSON, E. T. JR.
1910. Collecting and Mounting Micro-Diptera. Entomological News, 21: 406-410.

ESSIG, E. O.
1926. Insects of Western North America. MacMillan Company, New York, p. 907.

FELT, E. P.
1899. Collection, Preservation and Distribution of New York Insects. New York State Museum Bulletin 26: 14-25.
1929. A Popular Guide to the Study of Insects. New York State Museum Handbook 6: 18-101.
FORBES, W. T. M.
1915. Slides of Wings of Microlepidoptera. Journal of the Entomological Society of New York, 23: 69-70.
GARMAN, H.
1893. The Preservation of Larvae for Study. Insect Life, 6: 98.
GILLES, W. S.
1915. The Use of Formaldehyde in setting Insects. The Entomologist, 48: 41-42.
GUNDER, J. D.
1926. A New and Inexpensive Lining for Insect Boxes. Entomological News, 37: 152.
1926*a*. Butterfly Collections in Book Style. Entomological News, 37: 282.
HATCH, M. C.
1926. Concerning the Insect Collection. Entomological News, 37: 329-332.
HEATH, H.
1927. Caste formation in the Termite Genus Termopsis. Journal of Morphology, 43: 388-389.
HEGNER, R. W.
1916. Methods of Preserving Insects for Demonstration Purposes. Transactions of American Microscopical Society, 35: 185-186.
HOLBROOK, J. E. R.
1927. Apparatus and Method used to Remove Pins from Insect Specimens. Journal of Economic Entomology, 20: 642-643.
HOLLAND, W. J.
1922. The Capture, Preparation and Preservation of Specimens. The Butterfly Book, Doubleday, Page and Company, New York. pp. 26-57.
HUTCHINSON, C. M.
1925. Micro-anatomy of Cimbex with special Reference to Technique of Insect Histology. Indian Journal of Medical Research, Calcutta, 12: 487, 498.
JACKSON, A. D.
1907. Notes on Methods of Collecting and Preserving Thysanura. Ohio Naturalist, 7: 119-122.
KELLOG, V. L.
1908. Collecting and Rearing Insects. American Insects, Henry Holland Co., 657-661.
KELSHEIMER, E. G.
1928. The Preservation of Immature Insects. Annals of the Entomological Society of America, 21: 436-444.

KINGSBURY, B. F.; JOHANNSEN, O. A.
1927. Histological Technique. John Wiley and Sons, Inc., New York, 142 p.

LEE, A. B.
1928. The Microtomist's Vade-Mecum. P. Blakistons' Son and Company, Philadelphia, 710 pages.

METCALF, C. L.; FLINT, W. P.
1928. Destructive and Useful Insects. McGraw-Hill Book Company, Incorporated, New York, 229-297.

METCALF, C. L.; GLASGOW, R. D.
1922. The Insect Collection, Department of Entomology, University of Illinois. (Mimeographed directions, not published.)

METCALF, Z. P.
1926. A Killing Bottle for Collecting Small Insects. Entomological News, 37: 203-204.

MOSHER, F. H.; HOLBROOK, J. E. R.
1924. A Method of Inflating Larvae. Journal of Economic Entomology, New York, 17: 408-411.

MUSCHAMP, P. A. H.
1912. The Cleansing of Greasy Insects—A New Solvent. The Entomologist's Record and Journal of Variation, London, 24: 123.

PACKARD, A. S.
1873. Directions for Collecting and Preserving Insects. Bulletin Smithsonian Miscellaneous Collection 261.
1888. Entomology for Beginners, Henry Holt and Company, p. 294 ff.

RILEY, C. V.
1892. Directions for Collecting and Preserving Insects. U. S. National Museum Bulletin 39: 67-111.

SHELDIN, W. G.
1918. On a Cure for Insect Specimens Affected with Verdigris. The Entomologist, 5: 32.

SINTON, J. A.
1925. Notes on some Species of the Genus Phlebotomus (Dipt.) Journal of Medical Research 12: 801-806.

ST. JOHN, W. and Others.
1915. Formaldehyde Useful in Setting Insects. The Entomologist, 48: 42.

THOMPSON, F.
1914. Apparatus Designed for Setting Small Insects. Entomologist's Monthly Magazine, London, 25: 120-121.

TILLYARD, R. J.
1926. Collection, Preservation and Study of Insects. The Insects of Australia and New Zealand. Angus and Robertson, Sidney, Australia, 484-500.

TORRE-BUENA, J. R. de la.
1925. Methods of Collecting, Mounting and Preserving Hemiptera. Canadian Entomologist, 57: 53-57.

TOTHILL, J. D.
1919. The Chloral Hydrate Method of Preserving Insects for Dissection. Proceedings of the Entomological Society of Nova Scotia, Truro, 5: 8-10.

TOWER, W. L.
1903. Colors and Color Patterns of Coleoptera. University of Chicago Decennial Publication 10: 33-70.

WOMERSLY, H.
1927. Notes on the Mounting of Protura. Entomologist's Monthly Magazine, 3rd Ser., 63: 153-154.