PART B - CHAPTER 6

INDOOR SURFACE RESIDUE DISSIPATION

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GUIDELINE 875.2300

6.1 INTRODUCTION

This Guideline provides a description of the techniques and sampling strategies commonly used to characterize pesticide residues and dissipation on indoor surfaces. Such data are needed to quantify the transfer of dislodgeable residues from indoor surfaces to the skin. This information is used to determine whether or not a given pesticide may be used without appreciable risk in a residential setting.

The scope of the indoor environment encompasses a variety of settings in which human activities or properties are threatened by pests such as insects, microbes, rodents, fungi, or weeds. Examples of such areas where pest control is of interest are homes and apartments, greenhouses, farm buildings, health care facilities, schools and day care centers, and restaurants and food preparation establishments. Treated areas typically include the floors, carpets, furniture upholstery, cabinets, counter tops etc. of areas such as offices, kitchens, living rooms, and bedrooms.

Indoor surface residues may be generated from a variety of pesticide uses such as foggers, broadcast spray applications, crack and crevice treatments, vapor strips, moth repellents, residual termiticides, pet products, disinfectants, and indoor plant applications. According to the National Research Council (1993), the active ingredients found in most household products are cholinesterase-inhibiting compounds (i.e., organophosphates or carbamates). These chemicals can produce effects such as drooling and frequent urination, which may not be easily recognizable as resulting from pesticide intoxication because they resemble common behavioral patterns in children (Berteau et al., 1989). Other concerns include exposures to preservatives used indoors, such as those used to treat wood and those used in paint products. Outdoor-applied pesticides may also be tracked indoors where they become a secondary source of exposure (Nishioka et al., 1996). Also, persons who come into contact with pesticides as a result of their occupation may transport residues into their home (i.e., work clothing washed with other clothing, children touching contaminated clothing, etc.) Although EPA is cognizant of the existence of secondary exposure sources and the fact that humans, especially infants and children, are subject to these secondary sources, it recognizes that it is difficult to quantify these exposures due to limited data. Nevertheless, these secondary sources should not be overlooked when evaluating total human exposure.

Exposure to pesticides used in and around indoor and residential settings may occur via multiple routes of exposure -- dermal, inhalation, or nondietary ingestion. Dermal postapplication exposure results

when the skin contacts contaminated dust or surfaces, such as carpets, vinyl tile flooring, counter tops, upholstery, etc. (See Part B, Chapter 7 - Dermal Exposure.) Humans may be exposed to dust, vapors, and aerosols via the inhalation route. (See Part B, Chapter 8 - Inhalation Exposure Monitoring for techniques used to assess inhalation exposure, including indoor exposure.) Oral exposure (nondietary ingestion) may result from hand-to-mouth or object-to-mouth activity (especially for children), or through the consumption of contaminated food (including contamination while preparing, serving, and eating meals and snacks), and ingestion of dust or soil. (See Part B, Chapter 9 for information on non-dietary exposure assessment techniques.)

The techniques described in this chapter to measure indoor surface residues or dust residues may be used with the methods provided in Part B, Chapter 7 for dermal monitoring to estimate transfer of residues to human skin or with guidance provided in Part B, Chapter 9 - Nondietary Ingestion Exposure Assessment to estimate exposures from hand to mouth transfer.

The measurement of indoor pesticide residues is particularly important for populations that may spend a large portion of their time in indoor settings (i.e., children). Due to pesticide poisoning incidents involving children and the significant differences in the potential for pesticide exposures between adults and children, it may be necessary to focus specific attention on the assessment of pesticide exposures to children. The exposure potential for children (inclusive of infants and toddlers) may be greater than for adults because of several physiological, behavioral, and metabolic factors. The following are some examples:

- Children have a higher surface area to body weight ratio than adults.
- Although the inhalation rate in children is less than in adults, the volume inhaled on a body weight basis is greater in children.
- Children spend a significant amount of time crawling, playing or lying on the floor; consequently, pesticides may be absorbed by exposed skin if the contacted surface is contaminated.
- Children have increased mouthing activity and a lesser awareness of hygiene (i.e., eating food that has been dropped onto the floor, not washing hands after playing in dirt/soil), and as a result, it has been estimated that the risk of exposure to indoor and outdoor contaminants in soil and dust may be up to 12 times higher for children than adults (Hawley, 1985).
- The breathing zone for children is usually closer to the floor than adults.
- Infants may wear less clothing than adults while at home, i.e., wearing a diaper while crawling on the carpet, resulting in a greater surface area for potential exposure.

- Children, particularly infants, spend more time in the home than adults.

These factors along with other metabolic parameters and the stages of growth and development may make children more susceptible to passive indoor and residential exposures.

Recently, EPA has been conducting research aimed at developing methods for determining the concentrations of chemical substances on surfaces and in materials that are transferred to the skin or become available on the surface of the skin for possible dermal absorption and ingestion through hand-to-mouth activity (U.S. EPA, 1997a). The objectives of the research are to: (1) compare methods (i.e., wipe test, coupons, and dislodgeable residue) to determine the dissipation of malathion residues from residential media (carpet, vinyl flooring, and painted sheetrock); (2) measure the dissipation of residues of malathion and the production and dissipation of malaoxon under different conditions of humidity and temperature; and (3) determine the transfer of residues of malathion from the residential surfaces to human skin, cadaver skin, artificial skin, porcine skin, and passive dosimeter materials. The transferability of residues of malathion and malaoxon to human skin *in vivo* was compared to the transferability on human cadaver skin *in situ* and other skin substitutes. Transferability ratios were determined for the model skin surrogates. The results of this research are expected to assist in reducing the uncertainty associated with the relationships between pesticide residues in residential environments and the transfer of residues to the skin (U.S. EPA, 1997a).

6.2 SAMPLE COLLECTION

6.2.1 Test Substance

As stated at 40 CFR 158.390, the test substance to be used for inhalation exposure measurements must be a typical end-use product. Where metabolites, breakdown components, or contaminants of pesticide end-use products pose a potential toxicological concern, investigators may need to consider sampling for them on a case-by-case basis.

6.2.2 Timing of Application

Studies should be conducted under ambient conditions similar to those encountered during the intended use season or the season with the most frequent anticipated use. Ambient conditions (i.e., temperature, relative humidity, barometric pressure, ventilation, etc.) should be monitored throughout the study. Ventilation, among other factors, affects the accumulation, decay, transformation, transport (between rooms and media), and transferability (from media to body) of surface residues. Consequently, the time of application (e.g., summer versus winter) can impact the quantity of transferable indoor surface residues. For

instance, studies have demonstrated that relatively nonpersistent insecticides will remain within structures protected from sunlight and ventilation for several weeks (Leidy et al., 1993).

6.2.3 Pesticide Application Rate and Frequency

Generally, the typical end-use product chosen for the study should be applied at the maximum rate specified on the label. In addition to applying the product at the maximum label rate, it is suggested that the product be applied using a lower application rate, if possible. For example, typical rates are often used in cancer assessments (U.S. EPA, 1997b). Monitoring at more than one rate will provide additional information about the relationship between the application and deposition rates. Also, testing at a lower rate may prove to be beneficial in the event that the data from use of the product at the maximum application rate results in an unacceptable risk.

Where multiple applications are recommended, the minimum time interval between applications should be used. Also, the potential accumulation of residues from multiple applications should be considered.

Several methods are used in the application of indoor and residential pesticides. Some of the most common types of pesticides used indoors include: pesticide foggers such as flea bombs; spot or crack and crevice treatments; vapor strips for flying insects; broadcast spray applications; moth repellents; termiticides; pet products such as flea collars, dips and shampoos; disinfectants; and indoor plant applications. When several methods of application are available (e.g., fogger versus crack and crevice treatment), data should be provided in the protocol to support the rationale for selecting the application technique(s) monitored.

6.2.4 Sampling Parameters

Generally, indoor surface residue measurements should be collected over a sampling period that is sufficient to characterize residue dissipation. The following paragraphs provide a description of the locations where sampling should occur; how long the dissipation must be characterized (i.e., the sampling period); the times within the sampling period that samples should be taken (i.e., sampling intervals); and the number of samples that should be taken at each sampling interval along with a description of where at the sampling location to sample.

6.2.4.1 ***Number of Locations***

It is recommended that indoor surface residue (ISR) samples be collected from several different types of the media of interest. For purposes of an ISR study, the media of interest may be carpeting, hard surface flooring, counter tops, or other materials. It is recommended that as many types of the media of interest be sampled as possible to account for the broad variability in construction materials used. For example, if the media of interest is carpeting, the investigator may wish to sample a range of carpet types that represent differences in quality, stain resistance, and material. To ensure that the selected sample types are acceptable to the Agency, a study protocol should be submitted to the Agency before sampling begins.

6.2.4.2 ***Sampling Period***

Data should be collected in a manner that characterizes the dissipation mechanisms for the compound (e.g., three half-lives). Further, the sampling period should be reflective of the exposure conditions and toxicological endpoint of concern (i.e., acute or chronic). Typically, ISR dissipation rates are characterized for at least 72 hours postapplication unless the compound has been found to fully dissipate in less than this time period. EPA has found that this sampling period is adequate for characterizing pesticide dissipation under most use conditions; for most pesticides used in residential settings, significant dissipation occurs within the first 72 hours after application. Please note, however, that for more persistent pesticides a longer sampling period may need to be used. In addition, the recent passage of the Food Quality Protection Act may result in the need for monitoring over longer periods to allow for the aggregation of long-term exposures from multiple sources (i.e., dietary plus dermal). Registrants should present the proposed sampling period in the study protocol prior to initiation of the study to ensure that it is agreeable to the Agency.

6.2.4.3 ***Sampling Intervals***

Generally, the length of time between sampling should be relatively short in the beginning and should lengthen as the study progresses. EPA recommends that samples be collected prior to application on the day of application and on the application day at various intervals after application. For example, sampling at 1, 4, 8, 12, 24, 48, and 72 hours after application may be appropriate. These sampling intervals are provided for illustrative purposes only. Please note that for certain pesticides (e.g., one that degrades quickly) shorter sampling intervals may suffice. However, the sampling should continue for at least 72 hours, but all of the samples may not need to be analyzed if the pesticide has fully dissipated (i.e., nondetects for two sampling intervals). On the other hand, sampling may need to continue beyond 72 hours for more persistent pesticides. U.S. EPA (1996) observed detectable levels of chlorpyriphos residue in plush carpet up to 43 days after application. Transfer of chlorpyriphos residues to sampling media decreased by an order of magnitude within

the first month after application and by two orders of magnitude after three months. Special consideration should also be given to pesticides that exhibit biphasic dissipation kinetics. The data for the pesticide of interest should be collected for each of the selected surface media. The dissipation of the pesticide would be expected to be dependent on the types of surface media and the environmental conditions associated with its use.

The proposed sampling intervals should be presented to EPA for review in the study protocol prior to the initiation of the study to ensure that they are agreeable to the Agency.

6.2.4.4 *Number of Samples and Sampling Positions*

The number of samples and the sampling positions depends on the methodology selected (e.g., polyurethane foam roller). However, as a general rule, three samples per surface type per time period (i.e., triplicate samples) are recommended. The sampled areas should be marked so that they are not subsequently over-sampled. Frequently, a tape, ruler, or template is used to indicate where samples have been taken. Control plots should also be established. Sufficient control samples should be collected to ensure that the same bulk sample can be used as a negative control matrix throughout all sample analyses. Additionally, samples from the control plots should be collected at each interval for assessment of the field sample collection and storage procedures.

The areas chosen (e.g., rooms, hallways, etc.) should be representative of those typically treated with a pesticide and the environmental conditions expected in the intended use area. Variability in the surface types (e.g., stain-resistant carpet, hardwood flooring), surface conditions (e.g., old or worn vs. new), ventilation and air filtration, room size, etc. should also be considered.

6.2.5 Sampling Techniques

Several methodologies of assessing exposures to pesticides used indoors and around residential settings have been developed. Some of these techniques are used for measuring transferable surface residues from floors (e.g., Polyurethane Foam Roller, California Cloth Roller, and Dow Drag Sled) or other indoor surfaces (i.e., Lioy-Weisel-Wainman Sampler, Wipe Samples, and Hand Press). Others are used to sample bulk dust and debris (i.e., vacuum cleaners), or total residues (i.e., coupons). These guidelines will provide an overview of the current methodologies of measuring indoor pesticide total or transferable residues, as expressed in the published literature. Research is relatively new and is continuing in the area of indoor/residential exposure monitoring, and any guidance provided herein should be considered interim. Due to the lack of sufficient data to adequately endorse a specific sampling technique, EPA will not require the

performance of exposure studies using a specific technique, but a minimum acceptable criteria for conducting these studies will be provided. It will be at the discretion of the study investigator to select the methodology most suitable for measuring human exposure for the use(s) intended. In addition, the study investigator is encouraged to propose new methodologies to estimate human exposure and to validate existing methods. However, it should be noted that the selected technique must satisfy specific performance criteria as detailed in Part C, Quality Assurance/Quality Control.

The following list briefly describes some sampling methodologies currently described in the literature. For a more detailed explanation of the sampling methodologies, refer to "Methodologies for Assessing Residential Exposure to Pesticides" (U.S. EPA, 1994), and published literature. Recently, the efficiency of several of these methodologies (i.e., the polyurethane foam roller, California cloth roller, and Dow drag sled) for measuring dislodgeable residue transfer from floors (i.e., vinyl flooring and carpets) were compared (U.S. EPA, 1996). Information on the observed strengths and weaknesses associated with these methods, based on U.S. EPA (1996), are shown in Table 6-1. In addition, round-robin testing of several of these methods (i.e., the California cloth roller, the polyurethane foam roller, and the Dow drag sled) was performed to evaluate sampling precision (U.S. EPA , 1997c). The results indicated that reproducible and consistent data can be obtained using these methods (U.S. EPA, 1997c).

6.2.5.1 *Polyurethane Foam (PUF) Roller*

The polyurethane foam (PUF) roller sampler was designed to measure transferable residues from contaminated floor surfaces that a child may contact during various activities (i.e., crawling) (Lewis et al., 1994; Hsu et al., 1991). A dry PUF ring (8.9 cm outside diameter x 8 cm long) is secured on a 7.2 lb stainless steel roller. (See Figure B6-1.) The PUF ring is rolled over a surface once in both directions at the rate of 10 cm/second, exerting a pressure of 7,300 Pa, comparable to that of a toddler standing or crawling, (6,900 Pa - crawling and 8,600 Pa - standing). After the two rolls, the PUF roll cover is removed from the roller for analysis. The exposed rollers must be carefully handled to avoid contamination. Although it has been suggested that the PUF roller and other sampling devices be moistened with water to stimulate the moistness of the human skin, studies conducted by U.S. EPA (1996) indicate that the measurement variability increased substantially when moist contact media were used. According to U.S. EPA (1996), "the increased measurement variation with moistened media may be a serious impediment to the use of moistened media in field studies, since many more replicates would be required to detect a difference in transfer rates." Thus, the use of moistened media is not recommended unless first validated by the user for the specific pesticide formulation(s) under study. The EPA is continuing investigations to find a suitable moistening agent other than water. Nishioka et al.

Table 6-1. Observations from Field Use of Dislodgeable Residue Methods

Strengths	Weaknesses
Cloth Roller · Simple in design · Inexpensive to build from available materials	· Sampling cloth tends to bind and shift from original position · Plastic bag cover may adhere to PUF sleeve on roller from static · Difficult to operate due to mass of roller · Operator must contact treated surface · Susceptible to added pressure from operator · Transfer affected by roll orientation relative to lay of carpet fibers
Drag Sled · Simple in design · Inexpensive to build from available materials · Simple to use	· Drag contact unlike most skin contact with carpet · Drag contact is potentially directional relative to lay of carpet
PUF Roller · Consistent use across operators due to few variables · Relatively simple to use · Foam roller contact is more like skin contact	· Expensive to build or purchase

Source: U.S. EPA (1996)

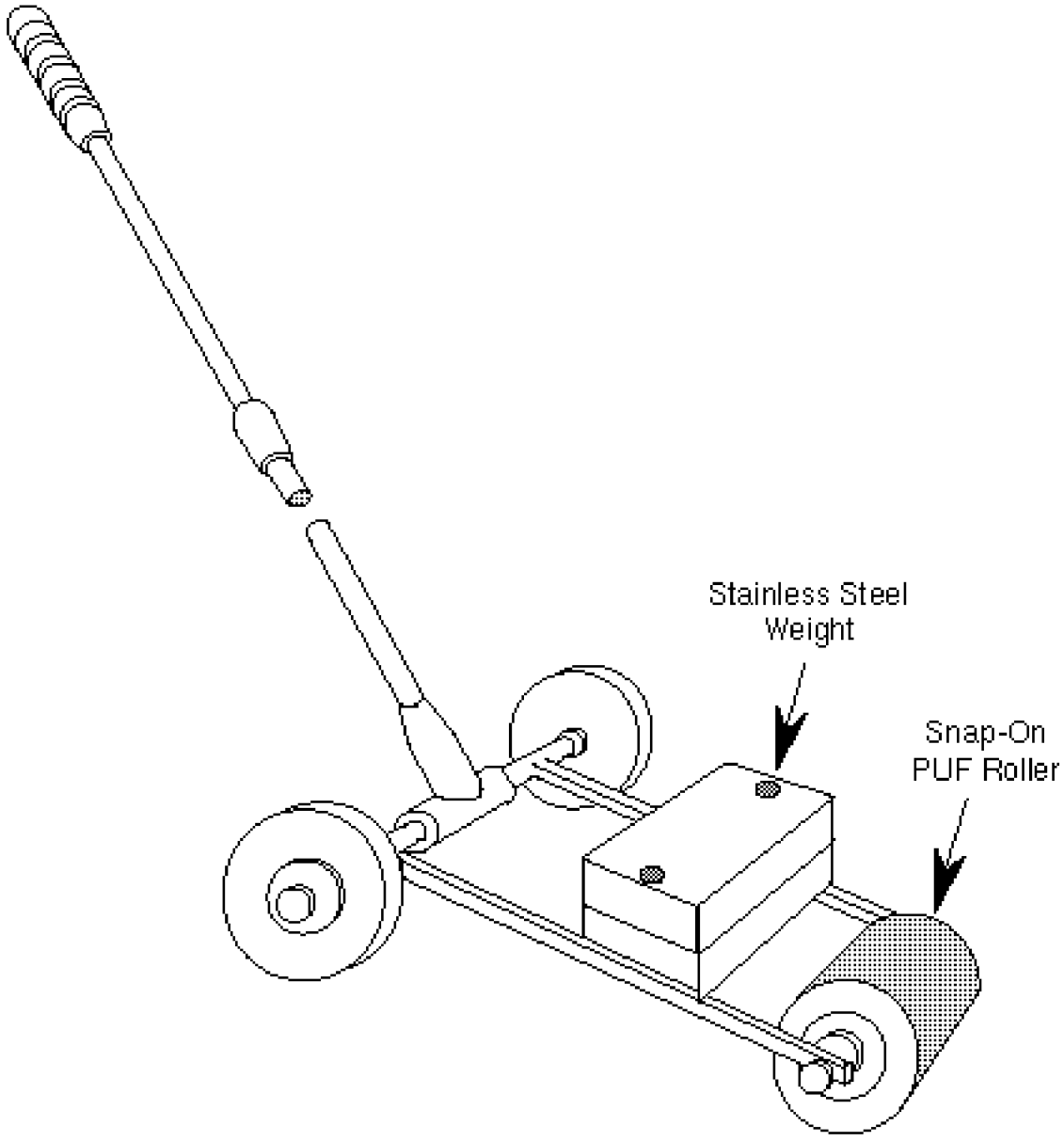
Stainless Steel
Weight
Snap-On
PUF Roller

Figure B6-1. PUF Roller Sampling Instrument

(1996) employed a "sweat simulator" composed of a buffered acetonitrile/water mixture to collect dislodgeable residues of several pesticides from carpet and turf.

6.2.5.2 *California Cloth Roller*

The California Cloth Roller technique is designed to collect transferable residues from floors. A percale cotton/polyester bedsheet is placed on the surface and covered with a sheet of plastic (or clean cotton toweling) (Ross et al., 1991). (See Figure B6-2.) A weighted foam covered roller (similar to a baker's rolling pin or paint roller) is rolled over the plastic bedsheet ten times backward and forward without additional pressure. After 10 passes, the percale cloth is collected, transported to the laboratory on ice, extracted, and analyzed. Recently, the Outdoor Residential Exposure Task Force (ORETF) developed a modification of the California Cloth Roller (Johnson, 1998). The roller is a 4-inch diameter by 24-inch long PVC pipe. The outside of the roller is wrapped with half-inch polyurethane foam sheet, or equivalent pipe insulation, for cushioning and traction. Enough weight is added to the inside of the roller to bring the total weight (excluding the handle) to 32 pounds, which is evenly distributed across the length of the roller. A handle is added to the roller. A rectangular-shaped frame is made of PVC material with inside dimensions of 24.5 x 36 inches. A 27 x 39 inch piece of 100 percent cotton cloth with a 200 thread count is secured to the frame with clamps. A piece of clear plastic, large enough to completely cover the cloth, is then placed completely over the cloth and also secured with the clamps. The frame assembly is placed in the plot with the cloth sampling media in contact with the turf. The frame is secured in place with spikes in each of the four corners of the frame to keep it from moving during sampling. The roller is placed just inside of the frame. Using the frame as a guide, the roller is pushed to the far end of the frame and pulled to the original starting point a total of five times. No downward pressure is exerted on the roller itself. The roller and frame are removed. Visible debris such as grass, thatch, granules, etc. are removed from the cloth sheet because this technique is designed to measure chemical residue which transfers to the cloth, not residue that adheres to particulate matter. The cloth is analyzed for chemical residue and the plastic is discarded. The roller and frame are reusable, provided they are decontaminated between sampling events. See Part B, Chapter 4 - Dislodgeable Foliar Residue Dissipation: Lawn and Turf, for more information. This modified method was developed for outdoor sites, but may also be suitable for use in indoor settings.

6.2.5.3 *Drag Samplers*

The Dow Drag Sled technique has been developed to estimate the transfer of pesticide from the contaminated floor surface to the skin (i.e., transferable residues) (Vaccaro et al., 1991). The technique consists of dragging a weighted (8 pound) 3 inch x 3 inch plywood block on which a removable denim

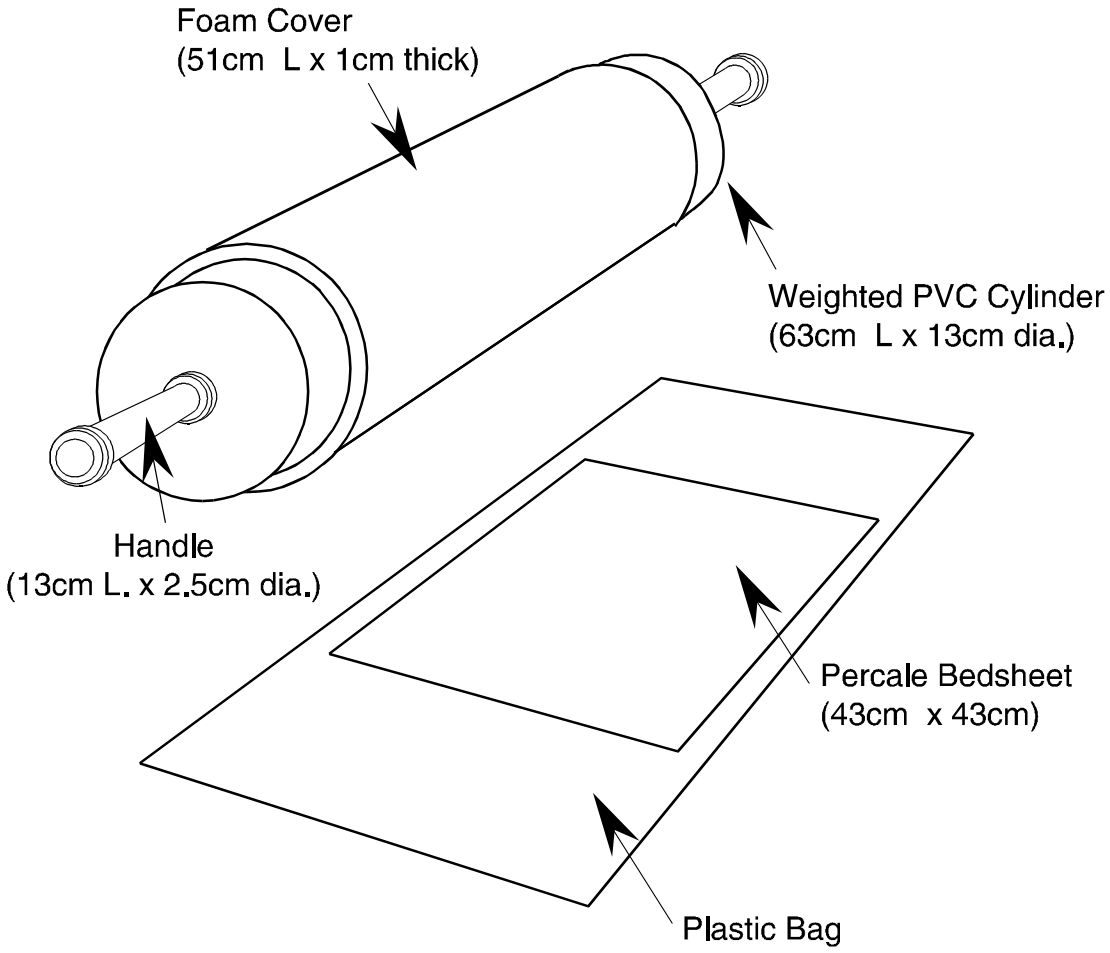

Figure B6-2. California Cloth Roller Sampling Device

patch is attached. (See Figure B6-3.) After dragging the sled once over a 3 inch x 4 foot carpet strip (sample area equal to 1-square foot) at 6-8 cm/second, the denim cloth is removed for analysis. The denim pad is removed after each drag.

Lioy et al. (1993) developed a similar device (i.e., the Lioy-Weisel-Wainman (LWW) Sampler) to sample flat surfaces. The sampler consists of two sections; one serves as a template and the other is a movable 4 cm square block that holds a filter sampling media that is moistened with "Type I reagent water." The template is placed on the sampling surface and the square block is dragged across the template opening, filter side down, a total of three times. Filters are then removed and analyzed for dust loading or the elemental content of sampled dust. Lioy et al. (1993) measured the reproducibility and dust (i.e., road dust and potting soil) collection efficiency of the LWW sampling device on three types of surfaces (i.e., painted shelving, formica, and wood paneling). For the purposes of these experiments, the filter media was polyethylene drain disc material that had been cut into rectangles. The results indicated "a high degree of reliability for flat surfaces." The authors noted, however, that porous surfaces may not be sampled as efficiently. Although Lioy et al. (1993) did not use the LWW sampler to sample for pesticides, the device may be suitable for measuring pesticides in surface dusts.

6.2.5.4 *Vacuum Cleaners*

Vacuum cleaners provide a method for collecting residues associated with dust and debris. In an effort to measure pesticides in house dusts, a potential reservoir or secondary source of exposure, a standard home vacuum cleaner has been used to collect samples from residential houses. In addition, a high volume surface sampler (HVS3) (Roberts et al., 1991a; Lewis et al., 1994) was designed to collect dust from carpets. HVS3 is a specially designed vacuum device with a stainless steel sampling train (Roberts et al., 1991b). (See Figure B6-4.) The device samples at a flow rate of 9.5 L/s (20 cfm) and passes the vacuumed dust through a cyclone that efficiently collects particles larger than 5 μm in a bottle at the base of the cyclone. A bulk sample of approximately 2 g of dust can be collected in up to 10 minutes. Studies employing a fine particle filter and PUF plug behind the cyclone have demonstrated greater than 99 percent of the vacuumed dust and 97 percent or more of tested pesticides (i.e., chlordane, aldrin, heptachlor, chlorpyrifos, and diazinon) are collected in the cyclone bottle (Roberts et al., 1991a; ASTM, 1996). Vacuum sampling devices are currently being used by EPA to collect dust samples for metals and pesticides analyses. It has also recently been used to collect carpet dust samples for pesticide analysis as part of the National Human Exposure Assessment Survey (NHEXAS) (Lebowitz et al., 1995). The HVS3 vacuum sampler has recently been used to collect carpet dust samples for pesticide analyses as part of the joint Agency (NCI, EPA, NIEHS) Agricultural Health Study (Camann et al., 1997). Concentrations of 32 pesticides in carpet dust from 9 farm households were evaluated.

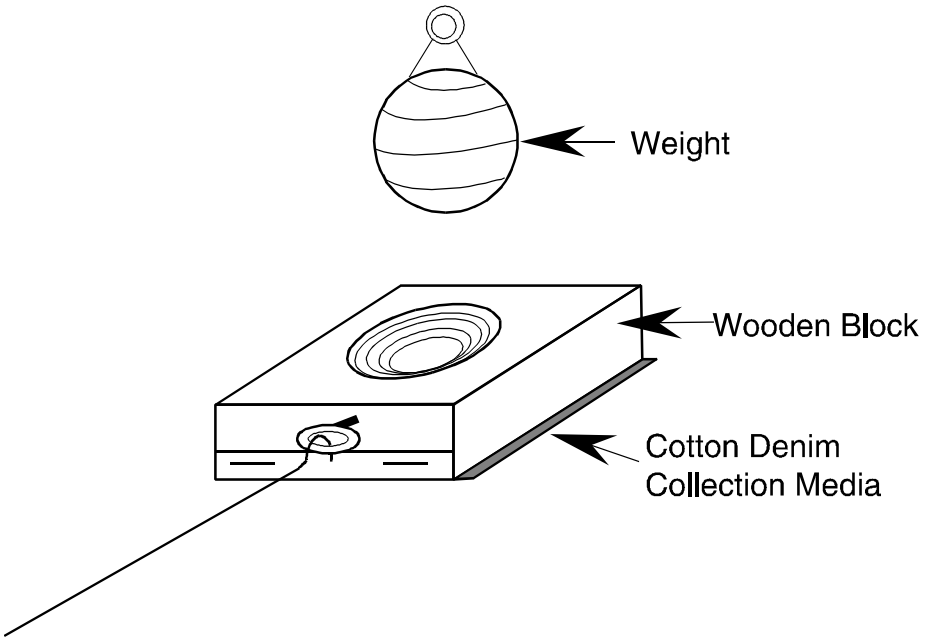

Figure B6-3. Dow Drag Sled Sampling Device

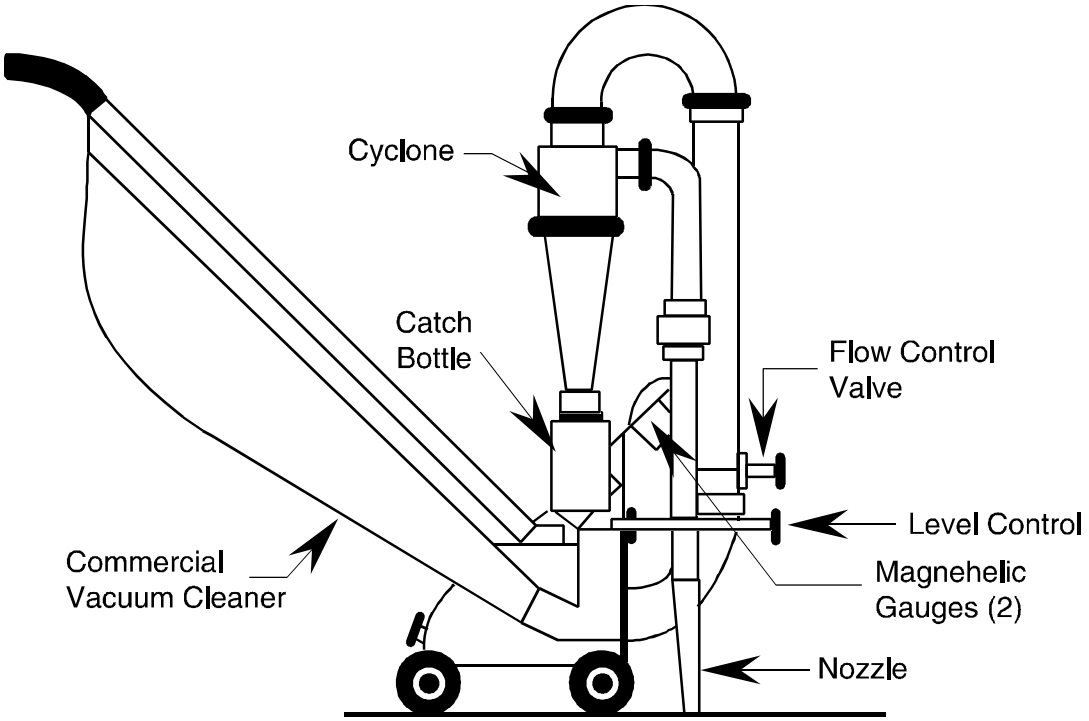

Figure B6-4. HVS3 Sampling Device

6.2.5.5 ***Coupons***

Coupons may be used to collect total or transferable residues. The "Gunther/Iwata" coupon approach is one method used for sampling the amount of total or transferable residues.

Typically, coupons are used to measure or confirm deposition rates (i.e., evaluation of the source strength). Deposition of pesticide residues may be measured using alpha cellulose or cotton gauze coupons placed in triplicate at indoor locations where deposition is likely to occur. The coupons are collected and residues are extracted with an appropriate solvent to measure total pesticide residues. Krieger et al. (1997) used 100 percent cotton dosimeters (i.e., T-shirt type material) to capture surface residues. The dosimeters were placed on aluminum foil to prevent penetration breakthrough, and positioned in locations on the floor where deposition was expected to occur. Under certain circumstances, it may also be appropriate to use the coupons consisting of the indoor material onto which the pesticide residues are deposited (i.e., textiles such as upholstery or carpet).

Coupons are also sometimes used to estimate dissipation rates. Coupons simulating materials commonly found in indoor settings (i.e., carpet, tile, hardwood, glass, etc.) are placed near or on the surfaces that they represent. It is essential that the coupon material used for such dissipation studies be representative of the indoor surface of interest. For example, carpet samples should not be used to represent hard surfaces. A sufficient number of coupons is placed at each location to provide triplicate samples for each sampling interval and field spikes. The coupons are collected at appropriate time intervals (i.e., before application, immediately after application, 2 hours, etc.), and an appropriate solvent is used to extract residues. Clean forceps should be used to pick up each coupon to prevent contamination between coupons.

6.2.5.6 ***Wipe Samples***

Residues that can be transferred from the treated surface as a result of contact can be measured using wipe sampling. This technique uses moistened cotton gauze pads to sample a standardized area (e.g., 1-square foot). This is a relatively simple technique that can be conducted on a variety of surfaces. The number of times that the surface should be wiped is not consistent. A weight may be attached to the sampling pad to apply uniform pressure. Research has shown that when two wipes are done (sampled area wiped twice using two pads in two directions and applying maximum pressure by the hand), the second wipe can yield almost as much residue as the first wipe (Naffziger et al., 1985). To minimize variability in results, certain factors should be considered (standardizing the sampling material, standardizing the area to be wiped, outlining the boundaries of the surface to be wiped with tape or a template, wiping the sample area once with firm even pressure, collecting samples in triplicate, checking the moisture content of the wipe). As with the

previous techniques, samples should be collected at sufficient intervals to establish a dissipation curve. In general, wipe samplers should only be used on hard surfaces (i.e., not carpeting).

6.2.5.7 *Hand Press*

The hand press is a sampling technique used to collect experimental data on transferable residues. It is typically used as an experimental research tool and not as a monitoring method. The hand press method is similar to the wipe sampling technique with the difference being the sampling medium (hands, with or without gloves, versus cotton gauze pads). Hsu et al. (1991) used the palm of the hand (excluding fingers) pressed at a pressure of approximately 1 lb/inch^2 sequentially over the designated testing area. This technique has also been used to validate the PUF roller technique (Hsu et al., 1991). In a technique developed by Lewis et al. (1994), the hand is first washed with soap and water and allowed to dry for 10 minutes. It is then pressed, palm-only, 10 times on a scale covered with cleaned aluminum foil to achieve a uniform pressure of 12 lbs. (5.4 kg, equivalent to 7,300 Pa), and then pressed, palm-only, 10 times sequentially on the carpet (or bare floor) along a 100 cm path using a 10 cm x 100 cm template.

6.2.5.8 *Migration Method*

Potential exposures to pesticides (e.g., antimicrobials) may occur as a result of residues that "leach" from treated articles (i.e., impregnated materials such as textiles or children's toys). Exposure to these residues may occur as a result of dermal contact or non-dietary contact (i.e., mouthing behavior). The U.S. Food and Drug Administration (FDA) recommends the use of a migration technique to assist petitioners in the preparation of the chemistry portion for indirect food additives (FDA, 1995). Indirect food additives are defined by FDA as "substances used in the processing, packaging, holding, and transporting of food that have no functional effect in the food but which may reasonably be expected to become components of food" (FDA, 1995). Although the Series 875, Group B Guidelines are not intended to address dietary exposures, this migration technique may have applicability in assessing migration of pesticides from impregnated materials to surfaces to which humans have non-dietary contact.

FDA (1995) recommends the use of a migration cell in which a material sample of known surface area is extracted by a known volume of solvent. FDA (1995) recommends that the surface area to solvent volume ratio should be reflective of the food and packaging materials of interest. The material sample is placed in the solvent so that the solvent flows freely around the material. "The headspace is gas-tight and liquid seals are maintained" (FDA, 1995). The cell is mildly agitated for 10 days or more at a temperature that is reflective of the temperature at which the material is used. The solvent is then analyzed for the chemical of interest. FDA (1995) recommends that the solvents used should simulate the food product that

will contact the impregnated material. For example, food oils may be appropriate simulants for fatty foods, and 10 percent ethanol may be used as an aqueous food simulant. For migration tests used for non-dietary pesticide exposure assessments, 10 percent ethanol may be an appropriate solvent. Recently, researchers have also been experimenting with artificial saliva and digestive extracts (personal communication between N. Freeman, EOSHI, Rutgers University, and L. Phillips, Versar, Inc., January 7, 1998) that may be appropriate solvents for extracting chemicals from impregnated products that may result in exposure from mouthing behavior. Methods have also been developed for modeling the flux from impregnated materials (U.S. EPA, 1992; U.S. EPA, 1997b). The model uses chemical-specific inputs such as molecular weight, material thickness, migration period, etc. to estimate migration from the material matrix to the surface. For more information on the migration estimation model, see Part D, Chapter 3 - Modeling.

6.2.6 General Considerations for Field Sample Collection

Surface sampling should be conducted in conjunction with air sampling. Enough air samples should be taken in each room to establish a dissipation curve. Duplicate samples at each time period is recommended. Stationary air samplers should be placed as applicable. Control (untreated) samples should be collected from the test site prior to application of the pesticide. Please refer to Part B, Chapter 8 - Inhalation Exposure, for more detailed information such as sampling intervals.

Control or background samples should be collected from the test site prior to application of the test substance. Sufficient control samples should be collected so that fortified controls can be prepared on each sampling day. These fortified controls should be packaged, transported, stored, and analyzed concurrent with the dislodgeable residue samples. (See Part C, Quality Assurance and Quality Control.)

6.3 SAMPLE STORAGE

Indoor surface residue samples and extracts should be stored in a manner that will minimize deterioration and loss of analyte between collection and analysis; more detailed information on sample storage is provided in Part C, Quality Assurance and Quality Control. The study investigator is responsible for demonstrating the stability of the samples under the storage duration and conditions used.

6.4 SAMPLE ANALYSIS

Dislodgeable pesticide residues should be collected from sampling materials (PUF, denim, etc.) as soon as possible. Validated methods of appropriate or sufficient sensitivity are needed for all sample

analyses. See Part C, Quality Assurance and Quality Control for more detailed information on sample analysis.

6.5 CALCULATIONS

Refer to Part D of this document for a description of the calculations needed for estimating dissipation rates, exposure, and risk.

6.6 DATA PRESENTATION

Indoor surface dislodgeable residues should be reported as mg or µg of pesticide active ingredient per m^2 (unit area) of surface area sampled. These data should be reported in tabular form for each sampling day. In addition, the best fit dissipation curve should be plotted (typically log-linear) with indoor surface dislodgeable residues on the Y-axis and time on the X-axis. Distributional data should be presented, to the extent possible.

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