DEPARTMENT OF HEALTH AND HUMAN SERVICES

Public Health Service
Centers for Disease Control
and Prevention (CDC)

Memorandum

Date: March 23, 2001

From: Roy M. Fleming, Sc.D., Director, Research Grants Program RMF
Office of Extramural Programs, NIOSH, D30

Subject: Final Report Submitted for Entry into NTIS for Grant 5 R01 OH003120-03.

To: William D. Bennett
Data Systems Team, Information Resources Branch, EID, NIOSH, P03/C18

The attached final report has been received from the principal investigator on the subject NIOSH grant. If this document is forwarded to the National Technical Information Service, please let us know when a document number is known so that we can inform anyone who inquires about this final report.

Any publications that are included with this report are highlighted on the list below.

Attachment
cc: Sherri Diana, EID, P03/C13

List of Publications

Tsai SW, Que Hee SS: A New Passive Sampler for Aldehydes. Am Ind Hyg Assoc J 60:463-473, 1999

Tsai SW, Que Hee SS: A New Passive Sampler for Regulated Workplace Aldehydes. Appl Occup Environ Hyg 14:255-262, 1999

Wiesenthal K, Jehlar A, Que Hee SS: Synthesis and HPLC/ultraviolet detection analysis of the 0-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine oximes of selected carbonyl compounds. J Assoc Offic Anal Chem Int, accepted, 1999

Shen Y, Que Hee SS: Optimization of a solid sorbent dynamic personal air sampling method for aldehydes. Appi Occup Env Hyg, in press, 1999

Title: Carbonyl Compounds Air Sampling Method
Investigator: Shane S. Que Hee, Ph.D.
Affiliation: University of California, Los Angeles
City & State: Los Angeles, CA
Telephone: (310) 206-7388
Award Number: 5 R01 OH003120-03
Start & End Date: 9/1/1995–8/31/1999
Total Project Cost: $515,055
Program Area: Exposure Assessment Methods
Key Words:

Abstract:
The objective was to develop workplace passive (diffusive) sampling and dynamic sampling methods for the regulated aldehydes and ketones for use in the workplace, both indoors and at hazardous waste sites. The specific aims were: (1) To synthesize pure O-(2,3,4,5,6-pentafluorobenzylamine hydrochloride) [PFBHA] derivatives of representative carbonyl compounds and quantify them using both gas chromatography and liquid chromatography; (2) To identify a generation of static (gas bag) and air dilution system (syringe pump air dilution system) with concentrations of hygienic interest for vapors of representative carbonyl compounds to demonstrate that the dynamic sampling method is quantitative at 0% and 90% relative humidity; and (3) To develop accurate diffusive sampling badges at 0% and 90% relative humidity that are sensitive for carbonyl carcinogens. Field studies were conducted on pathology laboratory personnel and workers exposed to formalin, glutaraldehyde, aldehydes and ketones.

This study found: (1) the PFBHA coated solid sorbent technique allow pg sensitivity with selectivity using GC/ECD or GC/MS-SIM for the aldehyde and ketone homologs. Less sensitivity was found with HPLC/UVD but both sets of O-oxime standards can be used for GC and HPLC if acetonitrile is the solvent. (2) Both the dynamic and passive sampling techniques for vapors of aldehydes and ketones show recoveries of at least 75% except for ketones that are sterically hindered by substitution at both β carbons to the carbonyl carbon, and those structures may effect these sites in the gas phase. (3) The field results of the passive and dynamic samplers appear to differ. When passive sampler was validated using a different technique the results are closer to the "correct" answer. More research is needed to further investigate aim 3. Passive sampling rates for ketones not completed in this study as well as defining the sampling rates using the new ACGIH ceiling limits. Analysis could also be initiated to determine sampling techniques using defined mixtures.

The extreme sensitivity of ECD for detection of PFBHA O-oximes of carbonyl compounds allows a high screening sensitivity that exceeds that for the standard EPA 2,4-dinitrophenyl hydrazine (DNPH) solid sorbent/HPLC-UVD method. Capillary GC allows far greater resolution than a C 18 reversed-phase HPLC column. While a nitrogen detector can be used for the sensitive screening detection of oxazolidine derivatives used for the current NIOSH and OSHA methods, PFBHA is more reactive than 2-(hydroxymethyl)piperidine and shows better recoveries for conjugated aldehydes like

acrolein and crotonaldehyde, and much better recoveries for ketones. The nitrogen detector for oxazolidines is more costly, and more expensive to run than a ECD detector.

GC/MS-SIM is far more selective (m/z 181) and sensitive for PFBHA O-oximes, than HPLC/MS as a confirmatory technique for 2,4-dinitrophenylhydrazones. GC/MS can be used to confirm oxazolidines for the NIOSH and OSHA methods, but the selectivity and sensitivity is worse.

The passive sampling configuration requires no pump calibration, and no investment in pumps as do dynamic sampling methods. Both passive and dynamic PFBHA samplers have a sampling rate for the aldehydes that is independent of temperature, RH, and intermittent sampling. There is a temperature dependence for the GMD passive sampler based on DNPH, and there is no 2-(hydroxymethyl)piperidine based passive sampler. In addition DNPH, being colored, is photochemically active in the ultraviolet/visible region of the spectrum. The PFBHA passive sampler is potentially patentable. Passive samplers are much more convenient to wear for the worker than the dynamic sampler-tubing-pump ensemble.

Passive sampling does not potentially upset the protectiveness of quantitatively fitted respirators under field conditions, unlike a dynamic sampler technique which might pull contaminated air through the cartridge or at the face to respirator seal. Since the breath is near 100% RH, and there is a temperature variation from workplace temperature to body temperature, it is advantageous to conduct studies of workplace protection factors (WPFS) for a badge that is not dependent on temperature and RH, unlike the GMD badge or charcoal-based diffusive samplers. Thus, at least for most carbonyl compounds, feasible measurements of WPFs are now possible.

Publications

Tsai SW, Que Hee SS: A New Passive Sampler for Aldehydes. Am Ind Hyg Assoc J 60:463-473, 1999

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FINAL PERFORMANCE REPORT

CARBONYL COMPOUNDS AIR SAMPLING METHOD

RO1 OH03120

November 30 1999

Principal Investigator: Shane S. Que Hee, Professor of Environmental Health Sciences

Co-Investigator: Weng Kee Wong, Associate Professor of Biostatistics

Department of Environmental Health Sciences
University of California at Los Angeles
School of Public Health
10833 Le Conte Avenue
Los Angeles CA 90095-1772
Phone: (310)206-7388
Fax: (310)794-2106
E-Mail: squehee@ucla.edu

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LIST OF ABBREVIATIONS

prefix M, mega-
prefix k, kilo-
prefix m, milli-
prefix μ, micro-
prefix, n, nano-
prefix p, pico-
°C, degrees Celsius
%, percent (v/v), (w/w), and (w/v)

ECD, electron capture detector
g, gram
GC, gas chromatography
HPLC, high performance liquid chromatography
L, liter
LC, liquid chromatography
m, meter
M, molar
MS, mass spectrometry
P, probability
PFBHA, O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride
ppb, parts per billion (v/v) and (w/v)
ppm, parts per million (v/v) and (w/v)
RH, relative humidity
SIM, selected ion monitoring
TIC, total ion current
UVD, ultraviolet detector

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ABSTRACT

The major aim of RO1 OH03120 was to develop dynamic and diffusive personal air sampling methods for the vapors of carbonyl compound liquids and gases that allow pg detection for use in the workplace, indoors, and at hazardous waste sites. The hypothesis was that PFBHA-based derivatization of carbonyl compound vapors and gases results in quantitative formation of O-oxime derivatives that can be uniquely detected at the pg level. The specific steps to prove the hypothesis were: (1) synthesis of pure PFBHA derivatives of representative carbonyl compounds, resolved uniquely by GC and by HPLC; (2)generation of static and air dilution system concentrations of hygienic interest for vapors of representative carbonyl compounds to demonstrate that the dynamic sampling method is quantitative at 0 and 90% RH; and (3) development of accurate diffusive sampling badges at 0 and 90% RH that are sensitive for carbonyl carcinogens. (4) Field studies.

Step (1) was possible for all 8 aldehydes studied which had hygienic guidelines (acetaldehyde; acrolein; chloroacetaldehyde; crotonaldehyde; formaldehyde; furfural; glutaraldehyde; and n-valeraldehyde), in addition to other aldehydes such as benzaldehyde; n-butyraldehyde; n-decanal; glyoxal; n-heptanal; isobutyraldehyde; and methyl glyoxal. The reaction of ketones showed a steric dependence. Ketones substituted at both β-carbons to the carbonyl carbon gave yields lower than 75%, and all others produced yields at or above 75%. Thus though diisobutylketone and 2,4-hexanedione gave poor yields, their O-oximes could still be synthesized. The 16 regulated ketones that produced O-oxime yields above 75% were: acetophenone; chloroacetone; 2-chloroacetophenone; cyclohexanone; diethyl ketone; diacetone alcohol; dipropyl ketone; ethyl amyl ketone; ethyl butyl ketone; methyl amyl ketone; methyl n-butyl ketone; methyl isobutyl ketone; 2-methylcyclohexanone; methyl ethyl ketone; methyl propyl ketone; methyl isopropyl ketone. In addition 2-heptanone and methyl glyoxal allowed yields >75%. A fine resolution temperature program allowed resolution of all aldehydes together or all ketones together by GC/ECD or GC/MS with least quantifiable limits being in the pg range. A completely new HPLC technique was developed that successfully resolved the O-oximes of 13 aldehydes and ketones.

Step (2) was investigated using the static Tedlar gas bag and the dynamic syringe pump air dilution techniques, and using a 10% PFBHA-coated Tenax TA pellet as a passive sampler, and 10% or 20% PFBHA-coated Tenax TA at 10 mL/min for the dynamic sampler using a 200 mg/ 100 mg front/back section configuration. All ketones and aldehydes that showed efficient O-oxime synthesis also allowed recoveries >75% on wet spiking in methanol at PEL-ppm-hr guidelines. The only exception was acetone which exceeded the capacity of the coated sorbent bed. All the aldehyde vapors were able to be generated near guideline levels and showed recoveries of >75% independent of temperature, RH, and intermittent flow. All the sterically unhindered ketones were similarly recovered for ketones with total exposures of ≤200 ppm-hr at 10 mL/min flow rate. Whereas a log linear relationship described the 75% recovery point at different PFBHA:carbonyl compound vapor molar ratios relative to flow rate for aldehydes, a log-log relationship was obeyed for ketone vapors so that the 75% recovery cut-offs for the molar

ratio of PFBHA:carbonyl compound vapor were more dependent on flow rate for ketone vapors than were those for aldehyde vapor. This was shown to be a kinetic effect. Thus of the ketones that allowed efficient wet chemistry synthesis of PFBHA O-oximes and efficient wet spiking, recoveries <75% were observed for vapors of acetophenone, ethyl amyl ketone, 2-chloroacetophenone, and acetone. The results for acetone were because the capacity of the sorbent was exceeded. The recovery for passive samplers exceeded 75% for cyclohexanone, diethyl ketone, ethyl n-butyl ketone, methyl n-amyl ketone, methyl n-amyl ketone, methyl n-butyl ketone, methyl isopropyl ketone, and methyl n-propyl ketone for wet spiking and vapor spiking. The sampling rates for these 8 ketones were calculated, and sampling rates for the other 8 regulated ketones could not be completed, but should present no problem. The adsorption behavior of representative aldehyde vapors was also studied to obtain insight into the rate determining step. The latter appears to be a hydrogen transfer or physical adsorption rather than the addition reaction itself.

Step (3) fulfillment at the pg level was possible for formaldehyde, acetaldehyde, crotonaldehyde, and furfural, the animal carcinogens, because of the sensitivity of GC/ECD and GC/MS-SIM, and the large number of the theoretical plates for the capillary GC column that allowed selectivity. Though the selectivity was also good for the C18 reversed phase column used for HPLC, pg sensitivity was not possible by use of ultraviolet detector, even at 200 nm. The setting of Ceiling limits for most aldehydes occurred during the grant funding period, and the methods developed here are more than adequate for OSHA PEL monitoring and most TLV monitoring as long as attention is paid to capacity, sensitivity, and selectivity.

Step (4) originally included two field sites, the UCLA work environment and a funeral mortuary. The contact at the mortuary was fired towards the end of 1997 and management did not want any sampling. Thus, it was decided to make do with sampling the UCLA campus work environment. The preliminary results show that workplace protection factors of quantitatively fitted respirators may not be adequate to protect workers who pour formalin and glutaraldehyde. Acetone pourers are not adversely exposed as long as they stay 3 foot upwind of the pour stream. Monitoring industrial hygienists should also wear respirator protection when monitoring at these formalin and glutaraldehyde pourings.

SIGNIFICANT FINDINGS

1. The PFBHA coated solid sorbent technique allows pg sensitivity with selectivity using GC/ECD or GC/MS-SIM for the aldehyde homologs, and for the ketone homologs. This fulfilled the major aim of the study. Less sensitivity was found with HPLC/UVD but both sets of O-oxime standards can be used for GC and HPLC if acetonitrile is the solvent

2. Both the dynamic sampling and passive sampling techniques for vapors of aldehydes and ketones show recoveries of at least 75% except for ketones that are sterically hindered by substitution at both β carbons to the carbonyl carbon, and those structure may affect these sites in the gas phase, for example, acetophenone, ethyl amyl ketone, and 2-chloroacetophenone even though adequate recovery was measured by synthesis of O-oxime in solution, and on wet spiking the ketone in methanol onto the coated solid sorbent. The proved the grant application hypothesis for all aldehydes and ketones, except for the exceptional ketones just mentioned.

3. The field results of the passive and dynamic samplers appear to differ. The passive sampler validated by an entirely different technique appears closer to the "correct" answer.

4. More work needs to be done to investigate the phenomenon in item 3, to complete the determination of passive sampling rates for the ketones that could not be completed, to inititate work with defined mixtures, and to define the sampling rates about the new ACGIH Ceiling limits.

USEFULNESS OF FINDINGS

1. The extreme sensitivity of ECD for detection of PFBHA O-oximes of carbonyl compounds allows a high screening sensitivity that exceeds that for the standard EPA 2,4-dinitrophenyl-hydrazine (DNPH) solid sorbent/HPLC-UVD method. Capillary GC allows far greater resolution than a C18 reversed-phase HPLC column. While a nitrogen detector can be used for the sensitive screening detection of oxazolidine derivatives used for the current NIOSH and OSHA methods, PFBHA is more reactive than 2-(hydroxymethyl)piperidine and shows better recoveries for conjugated aldehydes like acrolein and crotonaldehyde, and much better recoveries for ketones. The nitrogen detector for oxazolidines is more costly, and more expensive to run than a ECD detector.
2. GC/MS-SIM is far more selective (m/z 181) and sensitive for PFBHA O-oximes, than HPLC/MS as a confirmatory technique for 2,4-dinitrophenylhydrazones. GC/MS can be used to confirm oxazolidines for the NIOSH and OSHA methods, but the seelctivity and sensitivity is worse.
3. The passive sampling configuration requires no pump calibration, and no investment in pumps as do dynamic sampling methods. Both passive and dynamic PFBHA samplers has a sampling rate for the aldehydes that is independent of temperature, RH, and intermittent sampling. There is a temperature dependence for the GMD passive sampler based on DNPH,

and there is no 2-(hydroxymethyl)piperidine based passive sampler. In addition DNPH, being colored, is photochemically active in the ultraviolet/visible region of the spectrum. The PFBHA passive sampler is potentially patentable. Passive samplers are much more convenient to wear for the worker than the dynamic sampler-tubing-pump ensemble.

4. Passive sampling does not potentially upset the protectiveness of quantitatively fitted respirators under field conditions, unlike a dynamic sampler technique which might pull contaminat air through the cartridge or at the face to respirator seal. Since the breath is near 100% RH, and there is a temperature variation from workplace temperature to body temperature, it is advantageous to conduct studies of workplace protection factors (WPFS) for a badge that is not dependent on temperature and RH, unlike the GMD badge or charcoal-based diffusive samplers. Thus at least for most carbonyl compounds, feasible measurements of WPFs are now possible.

SCIENTIFIC REPORT

Background. The principal investigator (PI) in 1995 first reported (1) a personal dynamic solid sorbent method for aldehydes in air based on chemisorption of aldehydes (acetaldehyde, acrolein, crotonaldehyde, furfural, and valeraldehyde) with O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA). Tenax GC or Tenax TA were coated with 20% PFBHA. The method was not prone to effects of relative humidity (RH) as expected from the use of Tenax solid sorbents (2-4). Since there were no commercial O-oxime standards, wet spiking of the tubes with aldehydes was successfully compared with quantitations done with pure O-oximes synthesized by the PI (5,6). An earlier publication involved synthesis of the PFBHA O-oximes of formaldehyde, acetaldehyde, n-heptanal, n-decanal, and glyoxal for sampling of aldehydes in water (5). My group has also used the technique to quantify aldehydes and ketones in saliva from chewing tobaccoes (7).

The major aim was to develop workplace passive (diffusive) sampling and dynamic sampling methods for the regulated aldehydes and ketones for use in the workplace, indoors, and at hazardous waste sites. The accepted and submitted publications are provided in Appendices 1-8.

Appendix 1 contains the passive monitoring validation paper for 8 hr/day and 5 consecutive days/week sampling for n-valeraldehyde and acrolein, the aldehydes with the highest and lowest 1996 ACGIH TLV-TWAs, respectively (8). The sampler was a 10% PFBHA coated Tenax pellet (1.3 cm diameter) contained in a 3M 3500 organic vapor passive sampler (14,15) with a silicone membrane draft shield of diffusion path length 1.1 cm. Vapor and liquid spiking recoveries exceeded 91% and desorption efficiencies exceeded 96%. The critical face velocity was 15-20 ft/min. The capacity was about 30 μmol aldehyde. The sampling constants in the range 0.1-2 PEL equivalent were independent of intermittent sampling regime (continuous; high at beginning for 1 hr; high at end 1 hr; 3 exposure periods broken by periods of nonexposure), face velocities above critical, RH from 3-90%, and temperatures from 4 to 48 °C. Stability after sampling at 25 °C was at least 6 months. Shelf-life before sampling at 25 °C was at least 3 months. Both GC/ECD for screening and GC/MS for confirmation were possible with the same GC capillary column. The results were also compared with those when using the theoretical diffusion coefficients D calculated with the Fuller-Schettler-Giddings method (16).

Appendix 2 contains the extension of the passive sampling method to all OSHA regulated aldehydes (9), and those with ACGIH recommendations (acetaldehyde; chloroacetaldehyde; crotonaldehyde; formaldehyde; furfural; and glutaraldehyde). Each pure O-oxime was synthesized. Yields exceeded 82% (glutaraldehyde). The PFBHA dynamic air sampling method was first validated for each aldehyde using the gas bag technique as given for acrolein (1). The monoaldehydes all had recoveries exceeding 85% at 50 mL/min flow rate, but glutaraldehyde at this flow rate was 23% recovered. The dynamic sampling technique was then used to validate the passive sampling technique. The latter procedure was essential for low exposure concentrations. This phase resulted in successful aldehyde vapor phase generation using the syringe pump technique from sodium sulfite titrated aqueous solutions of the aldehydes provided commercially

for formaldehyde, crotonaldehyde, glutaraldehyde, and chloroacetaldehyde. The sampling constants for each aldehyde at PEL equivalent conditions were provided. All the aldehyde O-oximes were resolvable by GC/MS and GC/ECD. All vapor and liquid spiking recoveries of the passive sampler exceeded 85% relative to generated vapor mass balance. The capacity for the dialdehyde glutaraldehyde was half that of the monoaldehydes. The efficiency of sampling relative to the Fuller-Schettler-Giddings approach was within 25% for all aldehydes except chloroacetaldehyde where it was 1.6 times higher. The experimental D for acetaldehyde, formaldehyde, and glutaraldehyde agreed at $p \leq 0.05$ with those reported from the DNPH passive sampling method, except for acetaldehyde where the PFBHA-found D was 1.13 times higher.

Appendix 3 (10) contains the optimization and further validation of the dynamic sampling method with focus on valeraldehyde, acrolein, and formaldehyde, the latter because of its importance. Conditions for desorption were optimized. The dynamic sampling method recoveries were shown to be independent of RH, temperature, and intermittent exposure conditions. However there was a logarithmic-linear dependence of the PFBHA/ aldehyde ratio on sampling flow rate, that also implied passive sampling was possible.

Appendix 4 shows the extension of the optimized dynamic sampling method of Appendix 3 to regulated ketones (13). Many new PFBHA O-oximes had first to be synthesized for absolute quantitation. The ketones that showed vapor recoveries >75% at TLV-TWA equivalent conditions were: chloroacetone, cyclohexanone, diacetone alcohol, diethyl ketone, dipropyl ketone, ethyl butyl ketone, methyl amyl ketone, methyl butyl ketone, 2-methylcyclohexanone, methyl ethyl ketone, methyl isobutyl ketone, methyl isopropyl ketone, and methyl propyl ketone. Acetone showed breakthrough due to insufficient bed capacity. Acetophenone, 2-chloroacetophenone, ethyl amyl ketone, and diisopropyl ketone had recoveries <75% even though there was sufficient bed capacity, and the wet chemistry reactions showed yields >75%. This behavior was postulated to be because of steric effects at two carbons β- to the carbonyl carbon for the aliphatic aldehydes in the addition reaction (17,18) at the gas-solid surface, and the steric effect of an extra aromatic ring since adequate recovery was observed for benzaldehyde. There was a log-log relationship between recovery and ppm-hr exposure for chloroacetone, and between the PFBHA/aldehyde ratio and flow rate. There was a greater dependence of flow rate for ketones than for aldehydes. Ketones are the limiting factor when sampling mixed carbonyl compound atmospheres.

Appendix 5 contains the study to assess the influence of aldehyde and ketone structure on O-oxime synthesis, and to apply these derivatives for HPLC-UVD analysis (11). Seven new O-oximes were synthesized (acetaldehyde, acetophenone, benzaldehyde, n-butyraldehyde, n-decanal, methyl glyoxal, and n-heptanal) in addition to those for acetaldehyde, acrolein, crotonaldehyde, formaldehyde, glyoxal, and n-valeraldehyde. Isocratic and gradient elution reverse phase HPLC-UVD methods were developed that resolved all these O-oximes, including their E- and Z-isomers. Diisobutyl ketone and 2,4-hexanedione showed yields lower than 75%, unlike 2-heptanone, isobutyraldehyde, and 2-methylcyclohexanone. Inhibition occurred when both carbons β- to the carbonyl carbon were substituted. This implied a steric hindrance factor to

the addition reaction (17,18) to attain a tetrahedral intermediate. The detection limit was 100 ppb (w/v), with least quantifiable limits of about 500 ppb (w/v) at 200 nm. Thus GC/MS, GC/ECD, and HPLC-UVD methods can be used to analyze for O-oximes of aldehydes and ketones. Aldehydes and ketones in surface and tap water were determined at ppb (w/v) levels in 500 mL samples.

Appendix 6 shows the extension of the PFBHA passive sampling method of Appendices 1 and 2 to cyclohexanone, diethyl ketone, ethyl butyl ketone, methyl amyl ketone, methyl butyl ketone, methyl ethyl ketone, methyl isopropyl ketone, and methyl propyl ketone, using the dynamic air sampling method (Appendix 4) for validation. The sampling constants were calculated using the Fuller-Schettler-Giddings method (16). The experimental D values differed at $p \leq 0.05$ for all except diethyl ketone and methyl ethyl ketone. These results emphasized that experimental D have to be determined for accuracy, though the model gave D values within an order of magnitude of experimental D.

Appendix 7 contains the investigation of classical adsorption isotherms for the passive sampling studies on n-valeraldehyde and acrolein. All the classical adsorption isotherms (Langmuir, Brunauer-Emmett-Teller (BET), Freundlich, and Dubinin-Radushkevich (DB)) are obeyed. Their capacity terms and activation energies indicated the mechanism of the chemisorption of aldehydes at the gas/solid interface. The Langmuir and BET adsorption energies are characteristic of physical adsorption or hydrogen transfer. The DB limiting volumes are the largest and the BET/Langmuir capacities are the smallest. The rate determining step was either a physical adsorption or a hydrogen transfer, with two PFBHA sites occupied by an aldehyde molecule. This might account for the very different dependence on sampling flow rate observed in dynamic sampling due to a kinetic effect, and the lesser dependence found for passive sampling which is closer to thermodynamic equilibrium.

Appendix 8 shows the results of field studies to measure formaldehyde, glutaraldehyde and acetone personal breathing zone concentrations by dynamic and passive solid sorbent sampling based on O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride coated Tenax TA. The job practices investigated in a University job setting included pouring operations for dispensing and waste combination purposes, routine tissue fixing, and solvent cleaning purposes. A 11.7 eV photoionization detector and a dual flame ionization/10.6 eV photoionization instrument provided a collaborative analysis, in the "sniffing" mode. Acetone personal air concentrations were validated by parallel sampling with a charcoal tube, in addition to direct reading instruments calibrated to be direct reading to acetone. The workplace protection factors against formaldehyde and acetone for personnel who wore respirators are reported for the first time using passive samplers. Industrial hygienists who use direct reading instruments to monitor prolonged pouring and combination of formalin and glutaraldehyde for workers should also wear the appropriate respirator protection.

These papers show that the major air sampling and analysis methodologies for most regulated aldehydes and ketones for the PFBHA method have been established, and are practical under

OSHA guideline sampling conditions. Thus means that the grant application hypothesis has been proven. The PFBHA method has started to be applied by other investigators for air pollution uses (19-21). Other uses of PFBHA have been reviewed by the PI (6).

The specific aims of the grant application were:

(1) To synthesize pure PFBHA derivatives of representative carbonyl compounds (Appendices 2, 4, and 5 shows this) and to be able to quantify them by gas chromatography (all Appendices apart from Appendix 5) and liquid chromatography (Appendix 5).
(2) Generation of static (gas bag) and air dilution system (syringe pump air dilution system) concentrations of hygienic interest (OSHA and the ACGIH guidelines in 1994/95) for vapors of representative carbonyl compounds to demonstrate that the dynamic sampling method is quantitative at 0% and 90% relative humidity (RH). All appendices except Appendices 5 and 8 show fulfillment of this aim. The ACGIH guidelines have been much lowered since then, however.
(3) Development of accurate diffusive sampling badges at 0% RH and 90% RH that are sensitive for carbonyl carcinogens. Appendices 1 and 2 cover the passive sampling of the regulated aldehydes including the known animal carcinogens formaldehyde, acetaldehyde, furfural, and crotonaldehyde. However the ACGIH guidelines have been drastically lowered since 1994/95. Appendix 6 covers the passive sampling of the ketones. However there was no carcinogen amongst them and hygienic guidelines are usually of high concentration. The aldehyde technique is suitable for single ketones of TLV-TWA $\leq$25 ppm sampled over 8 hours and any with Ceiling Limits (chloroacetone was the only one in 1994/95).

Thus all of the then 8 regulated aldehydes can be sampled at 1994/95 ACGIH guidelines and 1999 OSHA PELs. Of the 29 regulated compounds in 1999 with keto groups and that have ACGIH guidelines, 16 solvent ketones were evaluated by dynamic sampling, and 8 regulated ketones have been validated for passive sampling relative to determination of sampling constant and synthesis of pure 0-oximes of PFBHA. Clearly, the rest of the sampling constants for passive sampling need to be done for the ketone solvents. The 7 solid regulated ketones (camphor, hydroquinone, isophorone, pindone, quinone, rotenone, and sucrose) and 2 other ketone gases (hexafluoroacetone and ketene) were not to be investigated, though in principle, their vapors should also be able to be detected and analyzed. The major ketone solvent omissions from the dynamic sampling ketones investigated are diisobutyl ketone, methyl isoamyl ketone, and methyl vinyl ketone. The latter had no guideline in 1994/95. The other two provide very low yields of O-oximes by wet chemistry, and represent the major non-specificity of the technique.

Field Studies. For the field studies (Appendix 8), collaborations occurred with UCLA Environment, Health and Safety (EHS) through Joseph Raab C.I.H., the UCLA industrial hygienist and Victor Kennedy C.I.H., the industrial hygienist of the UCLA Medical Center, for the following exposure situations:

(1) Assessment of formaldehyde exposure to pathology laboratory personnel, and those workers

who fill and empty formalin supplies and wastes. Most exposed personnel here are Caucasian women, with Black males dispensing formalin.
(2) Assessment of glutaraldehyde exposure from pouring operations. These operations are being phased out gradually because of the low ACGIH ceiling value. Most exposed personnel here are White women who are nurses or dental assistants. The dentists are usually male and of many ethnicities.
(3) Assessment of aldehyde and ketone exposures during bulking procedures at the UCLA central hazardous waste containerization facility before offsite treatment/storage/disposal. These EHS staff are White males.

The data of these field studies cannot be submitted for publication in peer-reviewed journals until the developmental papers still in review (Appendices 4 and 6) have been accepted. A draft of the not yet submitted paper is contained in Appendix 8.

ACKNOWLEDGMENTS

Please see the Acknowledgments sections of each Appendix. An extra thank you is for the collaboration of UCLA Environment Health & Safety in the field studies.

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8. SW Tsai, SS Que Hee, A New Passive Sampler for Aldehydes, Am Ind Hyg Assoc J , Accepted.

9. SW Tsai, SS Que Hee, A New Passive Sampler for Regulated Workplace Aldehydes, Appl Occup Environ Hyg, Accepted.
10. Y Shen, SS Que Hee, Optimization of a solid sorbent dynamic personal air sampling method for aldehydes, Appl Occup Env Hyg, Accepted.
11. K Wiesenthal, A Jehlar, SS Que Hee, Synthesis and HPLC/ultraviolet detection analysis of the O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine oximes of selected carbonyl compounds, J Assoc Offic Anal Chem Int, Accepted.
12. SW Tsai, SS Que Hee, A new passive sampler for regulated workplace ketones, Annal Occup Hyg, passed first review and in 2nd review.
13. YW Lin, SS Que Hee, A new dynamic sampling method for regulated workplace ketones, Annal Occup Hyg, passed first review and in 2nd review.
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PUBLICATIONS FROM RO1 OH03120

(i) Present:

1. SW Tsai, SS Que Hee, A New Passive Sampler for Aldehydes, Am Ind Hyg Assoc J , 60, 463-473, 1999. Ref 8. Appendix 1.
2. SW Tsai, SS Que Hee, A New Passive Sampler for Regulated Workplace Aldehydes, Appl Occup Environ Hyg, 14, 255-262, 1999. Ref. 9. Appendix 2.
3. Y Shen, SS Que Hee, Optimization of a solid sorbent dynamic personal air sampling method for aldehydes, Appl Occup Env Hyg, In Press. Ref. 10. Appendix 3.
4. K Wiesenthal, A Jehlar, SS Que Hee, Synthesis and HPLC/ultraviolet detection analysis of the O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine oximes of selected carbonyl compounds, J Assoc Offic Anal Chem Int, Accepted. Ref. 11. Appendix 5.

(ii) Anticipated:

a. In Current Review

1. YW Lin, SS Que Hee, A new dynamic sampling method for regulated workplace ketones, Appl Occup Environ Hyg, passed first review, and in 2nd review. Ref. 13. Appendix 4.
2. SW Tsai, SS Que Hee, A new passive sampler for regulated workplace ketones, Am Ind Hyg Assoc J, passed first review, and in 2nd review. Ref 12. Appendix 6.
3. SW Tsai, SS Que Hee, Vapor chemisorption classical isotherms of a solid sorbent passive sampler for aldehydes, Environ Sci Technol. Submitted. Appendix 7.

b. To be Submitted

1. S.S. Que Hee, Shih-Wei Tsai, Yang Shen, J Raab, V Kennedy. Aldehyde and ketone exposures for university workers. To Ann Occup Hyg. The developmental papers of a. 1 and 2 in current review must all be accepted before the field studies results will be submitted. Appendix 8.
2. Adsorption isotherm study for the dynamic sampling investigations (two papers). This will assess in depth the dependence of flow rate on the capacity and activation energy terms of the adsorption isotherms based on the modeling procedures of Appendix 7.

iii. **National Meeting Presentations**

1. Tsai, S.W., Que Hee, S.S., "Developing a New Aldehyde Passive Solid Sorbent Sampler", Am. Ind. Hyg. Conf. Expos., May 22-26, 1995, Kansas City, MO. Abstract 199.

2. Wiesenthal, K. and Que Hee, S.S. "Determination of Low Molecular Weight Aldehydes and Ketones by High Performance Liquid Chromatography", Am. Ind. Hyg. Conf. and Expos., May 17-23, 1997, Dallas, TX. Abstract 221.

3. Tso, J.C. and Que Hee, S.S., "Synthesis of Aldehyde Oxime Standards", Am. Ind. Hyg. Conf. and Expos., May 17-23, 1997, Dallas, TX. Abstract 222.

4. Tsai, S.W. and Que Hee, S.S. "Validation of a Passive Sampler for Valeraldehyde," Am. Ind. Hyg. Conf. and Expos., May 17-23, 1997, Dallas, TX. Abstract 195.

5. Tso, J-C. and Que Hee, S.S., "New Analytical Method for Determining Formaldehyde in Aqueous Samples", Am. Ind. Hyg. Conf. and Expos., May 11-15,1998, Atlanta, GA, Abstract 73.

6. Lin, Y-W. and Que Hee, S.S., "Validation of a Dynamic Personal Sampler for Chloroacetone", Am. Ind. Hyg. Conf. and Expos., May 11-15,1998, Atlanta, GA, Abstract 265.

7. Tsai, S.W. and Que Hee, S.S., "Validation of a Passive Sampler for Aldehydes", Am. Ind. Hyg. Conf. and Expos., May 11-15,1998, Atlanta, GA, Abstract 217.

8. Sheng, Y. and Que Hee, S.S., "Validation and Optimization of a Solid Sorbent Dynamic Personal Air Sampling Method for Aldehyde", Am. Ind. Hyg. Conf. and Expos., May 11-15,1998, Atlanta, GA, Abstract 211.

9. Sheng, Y. and Que Hee, S.S., "Sampling and Analysis of Airborne Environmental Aldehydes and Ketones", Am. Indust. Hyg. Conf. and Expos., Toronto, Ontario, Canada, June 5-11, 1999, Abstract 15.

10. Tso, J.-C. and Que Hee, S.S., "New Analytical Method for Determining Aldehydes in Aqueous Samples", Am. Indust. Hyg. Conf. and Expos., Toronto, Ontario, Canada, June 5-11, 1999, Abstract 16.

11. Tsai, S.-W. and Que Hee, S.S., "Validation of a Passive Sampler for Ketones", Am. Indust. Hyg. Conf. and Expos., Toronto, Ontario, Canada, June 5-11, 1999, Abstract 98.

LIST OF APPENDICES

Appendix 1. SW Tsai, SS Que Hee, A New Passive Sampler for Aldehydes, Am Ind Hyg Assoc J , 60, 463-473, 1999.
Appendix 2. SW Tsai, SS Que Hee, A New Passive Sampler for Regulated Workplace Aldehydes, Appl Occup Environ Hyg, 14, 255-262, 1999.
Appendix 3. Y Shen, SS Que Hee, Optimization of a solid sorbent dynamic personal air sampling method for aldehydes, Appl Occup Env Hyg, In Press.
Appendix 4. YW Lin, SS Que Hee, A new dynamic sampling method for regulated workplace ketones, Appl Occup Environ Hyg, In 2nd review.
Appendix 5. K Wiesenthal, A Jehlar, SS Que Hee, Synthesis and HPLC/ultraviolet detection analysis of the O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine oximes of selected carbonyl compounds, J Assoc Offic Anal Chem Int, Accepted.
Appendix 6. SW Tsai, SS Que Hee, A new passive sampler for regulated workplace ketones, Am Ind Hyg Assoc J, In 2nd review.
Appendix 7. SW Tsai, SS Que Hee, Vapor chemisorption classical isotherms of a solid sorbent passive sampler for aldehydes, Environ Sci Technol. Submitted.
Appendix 8. S.S. Que Hee, Shih-Wei Tsai, Yang Shen, J Raab, V Kennedy. Aldehyde and ketone exposures for university workers. To be submitted to Ann Occup Hyg.

APPENDIX 1

A New Passive Sampler for Aldehydes

AUTHORS
Shih-wei Tsai
Shane S. Que Hee*

Department of Environmental Health Sciences and UCLA Center for Occupational and Environmental Health, UCLA School of Public Health, 10833 Le Conte Avenue, Los Angeles, CA 90095-1772

A new solid sorbent passive air sampler used a coated Tenax TA pellet that chemisorbed aldehydes by reaction with 10% (w/w) O-(2,3,4,5,6-pentafluorobenzyl) hydroxylamine hydrochloride. The aldehyde permeated a silicone membrane to gain access to the sampling element at the end of a cylinder of diffusion path length 1.1 cm and diameter 1.3 cm. Vapors of known concentrations around the threshold limit values/time-weighted averages of n-valeraldehyde and acrolein and specific relative humidities (RH) were generated by syringe pumps in a dynamic generation and dilution system. An exposure chamber allowed measurement of face velocities, temperatures, exposing vapor concentrations, and RH. The O-oxime derivative was desorbed quantitatively with hexane, and an aliquot injected for gas chromatographic analysis on a low polarity capillary column using mass spectrometric or electron capture detection. The critical face velocity of the sampler was 15–20 ft/min and the capacity was about 30 μmoles. RH (3 ±1% to 79 ±2%) and temperature (4 ±1°C to 48 ±2°C) had no effects on the sampling constants of either aldehyde. Intermittent exposures had the same aldehyde equivalent recoveries as constant concentration exposures at the same time-weighted average. Stability after sampling was at least 6 months and the shelf life was over 3 months. The experimental sampling constants were 4.43 ±0.19 cm³/min for valeraldehyde and 7.73 ±0.57 cm³/min for acrolein.

Keywords: aldehyde, aldehyde oxime, adsorption, gas chromatography, passive sampler, personal sampling

Aldehydes (R-CHO where R is alkyl, aromatic, or alicyclic) are ubiquitous products of combustion,[1-6] water disinfection,[7] and biological oxidations,[8] and are mucous membrane irritants.[9] Formaldehyde, acetaldehyde, furfural, and crotonaldehyde are animal carcinogens.[10] Formaldehyde and glutaraldehyde expose embalmers,[11,12] operating theater personnel,[13] and pathologists.[9] The exposure and the health effects of formaldehyde have been extensively reviewed.[14,15] Methods for the analysis of aldehyde vapors usually use solid sorbent dynamic air sampling.[2,16-19] The advantages of passive samplers are well known.[20,21] The sampling constant k of a passive sampler is related to Fick's first law of diffusion as shown in Equation 1 in its form for a cylindrical open tube:[22]

$$dm/dt = (D_{AB}A/L)(c_{air} - c_{surf}) = k(c_{air} - c_{surf}) \quad (1)$$

where

dm/dt is the steady state mass sampling rate or mass transfer rate, weight/time

D_{AB} is the diffusion coefficient of the analyte B in air A, cm²/time

A is the effective cross-sectional area of the sampling element, cm²

L is the effective path length to the sampling element from the exposing atmosphere where diffusion control prevails, cm

c_{air} is the air concentration of the analyte, weight/cm³

c_{surf} is the air concentration of analyte just above the surface in the same units as c_{air}

k is the sampling constant of the analyte, equal to (DA/L), cm³/time

The dependence of the diffusion constant on molecular weight and temperature is expressed through Equation 2:[23]

*Author to whom correspondence should be addressed.

NIOSH/CDCP grant RO1 OH03120 and the UCLA Center for Occupational and Environmental Health were responsible for financial support.

$$D_{AB} = \frac{0.00143 \times T^{1.75}}{PM_{AB}^{1/2}\left[\left(\sum_V\right)_A^{1/3} + \left(\sum_V\right)_B^{1/3}\right]^2} \quad (2)$$

where

D_{AB} is the binary diffusion coefficient of analyte in air in cm^2/sec
T is temperature, K
M_A and M_B are molecular weight, g/mol
$M_{AB} = 2[(1/M_A) + (1/M_B)]^{-1}$
P is the external pressure, bar
Σ_v is the summation of atomic diffusion volumes, unitless
i is all the contributing species
A is air
B is analyte

Passive samplers have been developed for the lower molecular weight aldehydes and ketones based on liquid systems[24, 25] or solid sorbents coated with 2,4-dinitrophenylhydrazine (DNPH).[26-29] The DNPH method potentially allows specific quantitation of different aldehydes and ketones through high performance liquid chromatography (HPLC)/ultraviolet detection of their hydrazones but not by gas chromatography (GC) since many hydrazones decompose at high temperatures. The DNPH method does not react quantitatively with conjugated aliphatic aldehydes, can be light sensitive, and is of variable recovery on liquid aldehyde spiking.[2] Other coated sorbents have also been used for diffusive sampling of formaldehyde.[27,30]

Very sensitive GC/mass spectrometry (MS) and GC/electron capture detection (ECD) can be used to detect the O-oxime derivatives of O-(2,3,4,5,6-pentafluoro)benzylhydroxylamine hydrochloride (PFBHA) as reviewed elsewhere.[31] In the case of the reaction with PFBHA, each carbonyl group reacts with one molecule of PFBHA to form the corresponding O-oxime:[31]

$$\underset{\text{Carbonyl compound}}{R^1\text{–}(C{=}O)\text{–}R^2} + \underset{\text{PFBHA}}{R^3\text{–}O\text{–}NH_2\cdot HCl} \leftrightarrow \underset{\text{O-Oxime (E- and Z-forms)}}{R^3\text{–}O\text{–}N{=}C(R^2)\text{–}R^1} + H_2O + HCl \quad (3)$$

where R^1 and R^2 can be H-, aromatic, alkyl, or alicyclic functional groups and R^3 is the (2,3,4,5,6-pentafluorobenzyl) functional group.

Thus, the process involved here is chemisorption. For chemisorptive samples a reaction product is desorbed instead of the original analyte. The recovery of O-oxime in Reaction 3 above depends on two separate processes: the efficiency of reaction and the efficiency with which the O-oxime can be extracted for analysis (desorption efficiency). PFBHA-coated Tenax GC has been demonstrated to be an effective dynamic air personal sampling sorbent.[19] The latter is now shown to be an effective passive sampling system also.

MATERIALS AND METHODS

Materials

The aldehydes were acrolein (97%) and valeraldehyde (99%) from Aldrich (Milwaukee, Wis.) as was the GC internal standard, decafluorobiphenyl. Hexane (Optima), methanol (Optima), nitric acid, activated charcoal, molecular sieves, and indicating Drierite were from Fisher Scientific (Los Angeles, Calif.). PFBHA was from Lancaster Laboratories Inc. (Lancaster, Pa.). Tenax TA (80/100 mesh) was from Alltech Associates (Deerfield, Ill.). Helium, nitrogen, and 95% methane/argon were ultrapure grade from Alphagaz (Los Angeles, Calif.).

Equipment

The following were from Fisher Scientific: Pyrex® tubing 7-mm o.d. and 5-mm i.d. broken into lengths of 7 cm with their ends fire-polished in a propane-air flame of a piezoelectric microtorch (Blazer Products, New York, N.Y.); Pyrex glass wool cleaned by methanol and hexane Soxhlet-extractions; 4-mL Kimble vials with PTFE-lined screw caps (Fisher 03–340–60A); Pyrex ground glass volumetric flasks, beakers, V-vials, round-bottom flasks, spatulas, test tubes, and pipets; propipets; 10-μL Hamilton syringes; gastight Hamilton syringes; Soxhlet-extraction apparatus; bench centrifuges; Pasteur pipets; charcoal-lined respirator; vacuum desiccators; Buchi rotary evaporator; hot/cold air hair dryer; calibrated humidity/temperature meter/recorder; a Parr 2811 bench manual pellet press (Fisher Scientific 04–379); 3M Model 3500 OVM passive sampler; Bel-Art clear polycarbonate vacuum desiccator (Fisher Scientific 08–642–7) with a ceramic metal plate (Fisher 08–642–10); Harvard syringe pump (model 11), screw-caps 14-mm i.d. and depth 16 mm, a carbon dioxide incubator (Thermolyne Series 5000), heating tapes, Greenburg-Smith impingers; variacs, TFPE Teflon® and Tygon® tubing 6.4-mm i.d., and a Mettler AE260 analytical balance. Personal sampling pumps (model P30A) were from DuPont. Tedlar gas bags from 10- to 100-L were from SKC Inc. (Eighty-Four, Pa.). Stainless steel tubing of 6.4-mm o.d., stainless steel swagelok and ferrule T-adapters were from Alltech Inc. A Vibro-Graver vibrator was from Burgess Vibrocrafters (North Adams, Mass.). A M-5 Mini-Buck Calibrator for airflow rate calibrations was from Buck Scientific (East Norwalk, Conn.).

Solid sorbents were Soxhlet-extracted overnight with hexane and then methanol before being dried for 2 hours at 80°C in an incubator, with final drying to constant weight in a vacuum desiccator. Pure air was supplied by a Whatman Zero Air generator from Balston Inc. A 11.7 eV model PI-101 photoionization detector (PID) was from H-Nu, Inc. Rotameters were from SKC Inc. A calibrated model 8500 II hot-wire anemometer was from Alnor Instrument Co. (Skokie, Ill.). A small, box, desk fan for face velocity studies was part number 14244-01 from Tekna Design (Rockford, Mich.). A Goldstar Multiwave shelf microwave oven (Circuit City, Los Angeles, Calif.) facilitated PFBHA O-oxime syntheses.

GC/MS was done with a Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, Calif.) equipped with a 30 m × 0.32 mm i.d., 1 μm film DB-1701 chemically bonded fused-silica capillary column (J&W Scientific, Folsom, Calif.) linked with the 70 eV electron impaction source of a Hewlett-Packard 5988A mass spectrometer having a quadrupole mass filter and an electron multiplier detector. Injections of 2 μL hexane solution were at a helium carrier flow of 3.0 ±0.3 mL/min in the splitless mode with a purge delay of 1 minute. The temperature for the injector and link was 250°C. The column temperature program for valeraldehyde was solvent delay, 5 min at 105°C; 105°C for 0.5 min, 105°C to 180°C at 10°C/min, and holding then for 4 min. The column temperature program for acrolein was solvent delay, 5 min at 105°C; 105°C for 0.5 minutes, 105°C to 230°C at 10°C/min, and holding then for 9 min. The ion source temperature was 260°C. Selective ion monitoring utilized m/z 181 and total ion monitoring m/z 50 through 500. The areas of both

E- and Z- isomers were utilized for quantitations. Linear ranges were 200 to 1500 pg injected mass valeraldehyde equivalent and 180 to 3500 pg acrolein equivalent. Method detection limits (defined as the amount of analyte giving two times the background response) for valeraldehyde was 130 pg and for acrolein was 120 pg.

The same column and temperature conditions were used for Hewlett-Packard 5890 capillary GC/^{63}Ni ECD, with flows of 5% methane/argon being 3.0 ±0.4 mL/min, with a detector temperature of 250°C. The signal was visualized with a Hewlett-Packard 3396 integrator. Linear ranges were also 200 to 1500 pg injected mass for valeraldehyde equivalent and 180 to 3500 pg for acrolein equivalent injected mass in 2 µL injections. The detection limit for valeraldehyde was 110 pg and for acrolein was 100 pg.

Methods

Selection of Aldehydes

The two aldehydes selected for study represented the opposite extremes of the recommended 1996 threshold limit values/time-weighted averages (TLV®-TWAs) of aldehydes,[10] yet were non-carcinogenic, and were amenable to easy vapor generation. The lowest 1996 TLV-TWA was for acrolein (molecular weight 56.06 and boiling point 52.5°C) at 0.1 ppm or 0.23 mg/m^3,[10] and defined conditions for highest sensitivity. The highest TLV-TWA was for n-valeraldehyde (molecular weight 86.13 and boiling point 102°C) at 50 ppm or 176 mg/m^3 and defined the upper capacity required. Both were pure liquids and thus vapor generation through the syringe pump method was possible in a dynamic air dilution system. Both of the PFBHA O-oximes had been synthesized previously.[19] Aldehydes with TLV-TWAs (in ppm) between these two should behave similarly if the adsorption behavior of the two extreme compounds agree, assuming reaction efficiency is not affected with change in the R group. The dynamic sampling method has been validated for valeraldehyde and acrolein.[19]

Dimensions of the Passive Sampler

The sensing element was first made. PFBHA (300 mg) was dissolved in 25 mL of ASTM Type I water, and the solution added to 2.700 g of Tenax TA® in a weighed 250 mL 24/40 ground glass Pyrex round bottom. The water was removed by rotary evaporation at 85°C until a flowing solid occurred. The solid and container were placed in a vacuum desiccator containing indicating Drierite until a constant weight was achieved (usually after 3 days). A 150.0 ±1.0 mg weight of coated sorbent was then pelleted by a Parr 2811 bench handpress. The thickness of the pellet was 3 mm and the diameter was 13 mm.

For the initial studies a pellet was placed in the Teflon-lined screw cap of dimensions 18-mm o.d., 14-mm i.d., internal depth 14 mm, and outer height 16 mm (see Figure 1). The diffusion path length was 11.0 mm. In its field form the screw cap containing the pellet was secured to the bottom inner surface of the outer plastic casing of a 3M 3500 organic vapor monitor[32,33] after removal of the carbon cloth by duct tape. The Teflon stay of the 3M monitor was cut to allow the pellet to be held securely by one prong to maintain a constant diffusion path. The outer silicone membrane was then inserted on the top of the screw cap and fixed by an aluminum seal (Fisher Scientific 06-406-14B). The whole sampler was wrapped in aluminum foil (shiny side out) until sampling, which was initiated by removing the foil.

Exposure Chamber and Vapor Generator

Pure air for both the concentrated vapor stream and dilution air branches was generated by a Whatman Zero Air Generator using house compressed air (see Figure 2). It was still necessary to install leak-tight indicating Drierite and charcoal canisters after the generator to indicate when the purified air was sufficiently dry and free of organic vapor for use. After 30 min at an input air pressure of 75 psi, the total hydrocarbon concentration was <0.1 ppm and the RH was <2.0%. One calibrated rotameter was in each branch of the generation system before entry of any organic vapor or humidity. The range for each rotameter was 0.46 to 4.6 L/min

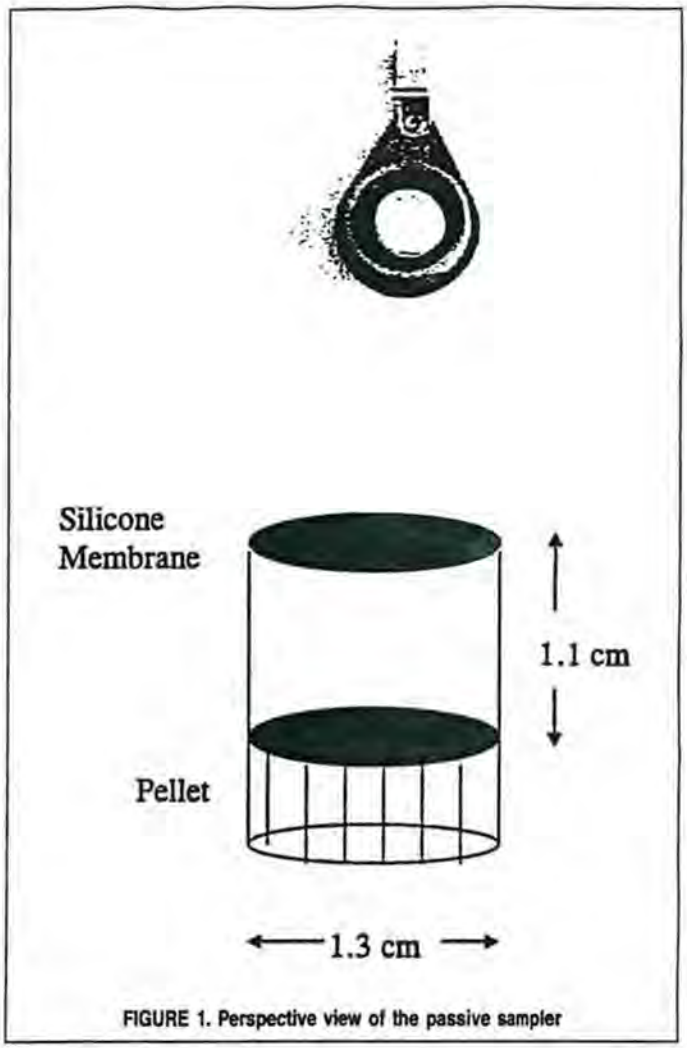

FIGURE 1. Perspective view of the passive sampler

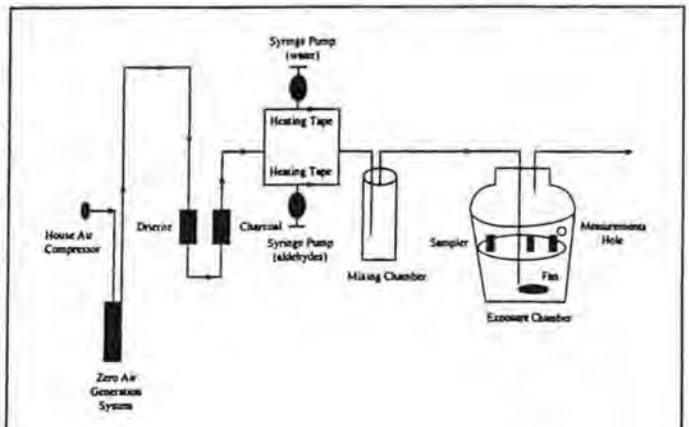

FIGURE 2. Aldehyde vapor generation and exposure chamber system

for the air dilution branch and 0.1 to 1.5 L/min for the concentrated organic vapor air stream. The air generator was connected to the vapor and water generation sites by 0.25 inch × 0.21 inch i.d. stainless steel tubing (type 316). The generators were syringe pumps set at known plunger velocities to generate the desired concentration of organic vapor for dilution, or RH for humidification. Heating tape wrapped around the outside of the stainless steel tubing at the needle exit from the syringe pumps ensured total volatilization of organic vapor or water. The two streams were then routed through a stainless steel T-joint adapter, and the outlet connected by Teflon tubing of 6.4-mm i.d secured by butt-to-butt joints with Tygon collars to a 1-L Greenburg-Smith impinger, which acted as a mixing chamber. The entire air generation and dilution system was mounted on a movable two-tiered laboratory cart.

Teflon tubing then conveyed the diluted organic vapor into the exposure chamber through a hole bored on the side near the chamber bottom to just underneath the fan blades, the fan resting at the bottom of the chamber and under the 23-cm diameter ceramic metal plate containing 11-mm diameter holes and a center 30-mm hole. The height of the exposure chamber/desiccator was 33 cm and the outer diameter was 28 cm. Remote control with a variac allowed different fan blade velocities and hence face velocities, as well as adequate mixing as shown by direct reading organic vapor analysis and hot-wire anemometry. Six samplers were set horizontally on the plate, each with a nearby closable hole in the chamber wall for probe insertion for measurement of RH, temperature, organic vapor concentration, and face velocity. Teflon tubing vented excess vapor to a fume hood. Direct reading confirmation of the diluted organic vapor concentration was possible only for valeraldehyde. The PID was calibrated by the static method using a measured volume of aldehyde injected into a known volume of air,[19,34] and the bag was heated briefly with a hot-air hair dryer. Acrolein concentrations in the exposure chamber were assessed by the published dynamic PFBHA method[19] involving a personal sampling pump set at a known flow rate, desorption of the O-oxime, and subsequent GC/MS or GC/ECD quantitation using the internal standard method. The whole system was made leakproof through assessment by a soap bubble solution, and subsequent appropriate tightening and application of Teflon tape. All tubing was hard-wired with nichrome wire.

Validation Protocol

The overall validation protocol is summarized in Table I. Each step was done at least in triplicate, and in the following order.

Synthesis of PFBHA O-Oximes

The acrolein and valeraldehyde PFBHA O-oximes were first synthesized by methods detailed elsewhere.[19] The respective GC/MS purity and yield percentages were 97.7 ±2.3 and 90.0 ±2.0 for acrolein, and 99.0 ±0.9 and 90.6 ±2.9 for valeraldehyde. The respective GC/ECD purity percentages were 98.2 ±1.7 for acrolein and 96.5 ±2.5 for valeraldehyde.

Desorption Efficiency

A volume of 50 μL hexane solution containing the theoretical amount formed after sampling 8 hr of exposure at the TLV-TWA was spiked onto the pellet. The spiked pellet was held overnight in a desiccator containing Drierite to allow the hexane to dry before desorption with 2.0 mL hexane over 2 hours, 30 sec of ultrasonication, with standing at room temperature over 2 hours before analysis by GC/MS or GC/ECD.

Reaction Efficiency/O-Oxime Recovery for Wet Spiking of Aldehyde

A weight of 1.338 mg of liquid valeraldehyde (two times the TLV 8-hr mass) was spiked onto the sampling pellet. Some of the spiked pellets were (1) kept at room temperature after spiking for 5 days; (2) held at 4°C for 2 days and then at 25°C for 3 days; (3) held at 4°C for 5 days; and (4) held at 4°C for 5 days and desorbed at 4°C. GC/MS and GC/ECD analyses were then performed as detailed for the desorption of the PFBHA O-oximes. The same procedure was used for acrolein except it was spiked at 1.81 μg (2 TLV × 8 hr) in 50 μL methanol.

Face Velocity

Starvation effects at low face velocities are expected to be important in chemisorptive samplers as for samplers that operate through physical adsorption. Thus, the critical face velocity of the system had to be defined. The presence or absence of a membrane to define a diffusion path length L will also affect the critical face velocity unless the sampling element sits at the end of a long thin tube beyond the critical L. The latter design is a potential safety hazard to wearers, so a shallow wide-mouth sampler design was favored. A membrane also prevents aerosol contamination of the collection element and defines the sampling as gas/vapor only. The critical face velocity must be exceeded to have valid sampling constants.

Face velocity effects were evaluated by exposing the sampler to the mass equivalent to the TLV-TWA vapor concentration over 8 hr at face velocities between 0 through 70 ft/min at intervals of 10 ft/min. A calibrated hot-wire anemometer was used to measure the face velocity just above the pellet surface, or if a silicone membrane was used, just near the latter's surface. The mass desorbed by the method used for the wet aldehyde spiking method was then divided by the mass that should be sampled using the sampling constants at the appropriate temperature in Table II. This multiplied by 100 was the sampling efficiency relative to the theoretical from Equation 2 and was plotted for each face velocity against the arithmetic mean of the measured face velocity. The critical face velocity was determined when the sampling rate decreased to the 95% confidence level of the steady state sampling rate.[21]

Vapor Exposures

All of the exposures in Table I were performed at 25 ±1°C, 36 ±2 % RH, and above the critical face velocity. For valeraldehyde, the calibrated PID monitored the chamber vapor concentrations in Table I. The total ppm-hours was obtained by summing the area under the parts per million versus time exposure plots. This was not possible for acrolein since no direct reading instrument was sensitive enough. Instead, the dynamic sampling method at 50 mL/min flow rate was utilized to make a direct integrated and simultaneous comparison (the dynamic sampling efficiency was 94 ±6%).[19]

In a typical run for valeraldehyde (density 0.818 g/mL at 11°C), the syringe pump was set at 0.200 mL/hour into an airflow rate of 829 ±17 mL/min diluted to a total flow rate of 2921 ±44 mL/min for exposure of the passive sampler for 70 min at a theoretical air concentration of 275 ppm or 0.80 the TLV × 8-hr dose. The actual exposure dose as monitored through the calibrated PID was about 300 ppm-hours taking the rectangular hyperbola exposure shape into account or about 0.75 of the TLV × 8-hr dose after correction to 25°C and 760 mm Hg. The usual protocol involved generating 0, 0.25, 0.5, 1.0, 1.5, and 2.0 times the TLV × 8-hr vapor doses over 1 hr of exposure.

TABLE I. Parameters to be Evaluated in Order of Performance for Chemisorptive Passive Sampling Evaluation for Acrolein (1996 TLV-TWA 0.1 ppm) and n-Valeraldehyde (1996 TLV-TWA 50 ppm) Using GC/MS and GC/ECD Analysis for PFBHA O-oximes

Parameter	Conditions
Aldehyde markers	Identification and synthesis for standardizations
Desorption efficiency of O-oximes	O-oxime mass equivalent to air concentration (ppm) × 8-hour at theoretical sampling rate[A] at concentration 0 and 1.0 times the TLV-TWA; wet and dry conditions relative to hexane solvent
Reaction efficiency of aldehyde plus desorption efficiency of O-oxime; wet spiking	Aldehyde mass equivalent to air concentration (ppm) × 8 hr at theoretical sampling rate[A] at concentration 0 and 2.0 times the TLV-TWA; wet and dry conditions relative to 4°C and 25°C; desorption with hexane; methanol (50 μL) was solvent if volume of liquid aldehyde <1 μL
Face velocity	0, 10, 20, 30, 40, 50, 60, 70 ft/min at (36 ± 2%) RH and (25 ± 1)°C at ppm-hr for TLV; hot-wire anemometer probe must be calibrated at these conditions; critical face velocity determination; with and without membrane
Vapor exposure	At 25°C for defined RH and above critical face velocity; effects of reverse diffusion on marker; ppm-hr equivalent generation to TLV × 8 hr in five protocols: (i) continuous exposure at TLV for 8 hr; (ii) at TLV-8 hour equivalents from 0–1 hr followed by zero generated air from 1–5 hr; (iii) at zero generated air from 0–3.5 hr with TLV-8 hr equivalents from 3.5 to 5.0 hr; (iv) at TLV-8 hour equivalents from 0–1 hr; (v) at TLV-8 hour equivalents for generation hours 0–1, 1.5–2.5, and 3.8 to 5.0 with zero air generated in between
Sampling constant	At 25°C for defined RH and above critical face velocity involving vapor concentrations of aldehyde at ppm-hr equivalent to 0, 0.1, 0.5, 1.0, and 2.0 times the TLV for 8 hr for field version and sensing element over 1-hr exposures
Capacity	Vapor and aldehyde liquid spiking exposures
Relative humidity	3 ± 1%, 36 ± 2%, and 79 ± 1% at 25.0 ± 1.0°C at the ppm-hr for the TLV over 8 hr
Temperature effect on sampling constant	9 ± 1°C, 25 ± 1°C, and 48 ± 2°C at specific RH and at the ppm-hr for 0.5, 1.0, and 2 times the TLV over 8 hr
Storage stability	4 and 25°C weekly over 6 months for samplers exposed to known vapor concenrations equivalent to the TLV for 8 hr
Shelf life	25°C storage over 3 months analyzed weekly to assess whether O-oxime produced relative to same vapor concentration challenge over the same time varies from when pellet was just made

[A] Using Equations 1 and 2.

TABLE II. Theoretical Sampling Constants at Different Temperatures from Equation 2

	Valeraldehyde		Acrolein	
Temp.[A] (°C)	Diffusion Coefficient (cm^2/sec)	Sampling Constant[B] (cm^3/min)	Diffusion Coefficient (cm^2/sec)	Sampling Constant (cm^3/min)
8	0.074	5.37	0.098	7.11
25	0.083	5.95	0.108	7.84
48	0.093	6.75	0.123	8.93

[A] Temp is the temperature.
[B] Sampling constant k was calculated from Equation 1 with C_{out} assumed equal to zero.

For zero (<1 ft/min) face velocity experiments, the chamber was allowed to come to concentration steady state before inserting the six samplers. On attaining steady state again (8 to 11 min depending on concentration), all exits were closed and made leak-proof with the inside concentration monitored with a direct reading instrument for valeraldehyde or low flow (50 mL/min) pump for acrolein for 70 min before O-oxime desorption and GC/MS and GC/ECD analysis.

Capacity

All of the capacity experiments were done at 25 ±1°C. Both wet aldehyde and aldehyde vapor capacities were determined using hexane for desorption, and GC/MS or GC/ECD for analysis. The vapor studies utilized high vapor concentrations to assess the asymptote in O-oxime mass desorbed. The procedure for wet spiking is given elsewhere.[19]

Relative Humidity Effects

Since water is a product of the reaction, there may be low reaction efficiency in humid air. The aqueous derivatization in excess PFBHA proceeds quantitatively on heating.[35] Acid also catalyzes Reaction 3.[35] Thus, potentially, Reaction 3 on a solid sorbent should be as efficient as the aqueous reaction, and this indeed was demonstrated for the dynamic air personal sampling method for the aldehyde vapors and liquids investigated after flow rates were optimized.[18] The independence of Tenax GC to RH effects for the PFBHA-coated sorbent has already been documented,[19] as has the small effect of RH on retention volumes of adsorbates.[36] Both aldehydes (including acrolein and acetaldehyde), and ketones are retained on uncoated Tenax GC.[37] This may allow further opportunity for reaction with PFBHA molecules at the coated sorbent surface. Tenax TA, the replacement for Tenax GC, has similar properties.[38]

Three different RH atmospheres (3 ±1%, 36 ±2%, and 79 ±2%) were cogenerated along with the aldehyde vapor at the same exposure and analysis conditions for the sampling constant.

Temperature

Decreasing temperature increases physical adsorption efficiency.[21] Diffusion coefficients usually increase to the absolute temperature raised to the 1.5 power (Equation 2). The air concentration a_c varies inversely with absolute temperature according to the ideal gas law in Equation 4:

$$a_c = m/V = pM/RT \quad (4)$$

where

m is the mass of analyte, g
V is the volume of air, L
p is the partial pressure of analyte, atmos
M is the molecular weight of analyte, g/mol
R is the gas constant in appropriate units, 0.08205 L atmos^{-1} K^{-1} mol^{-1}
T is temperature, K

The net result is a square root direct dependence on absolute temperature for the vapor sampling part of physical adsorption without considering the condensation behavior of the analyte.

For chemisorption, reaction rate may have a low or high temperature coefficient depending on distance apart of the reactant molecules on the surface.[39] Reaction rate increases exponentially with absolute temperature for uncatalyzed reactions obeying an Arrhenius law (Equation 5):

$$k = A\, e^{-E/RT} \quad (5)$$

where

k is the first order rate constant for the reaction or process, time^{-1}
A is the Arrhenius orientation or frequency factor, time^{-1}
E is the activation energy, cal/mole
R is the gas constant, cal K^{-1} mol^{-1}
T is the absolute temperature, K

However, for excess PFBHA and since PFBHA is also acidic in its hydrochloride salt form constituting a catalyst, there may be a small temperature coefficient over the limited temperature range usually recommended[21] in the National Institute for Occupational Safety and Health (NIOSH) protocol of 10 to 40°C. Clearly the temperature dependence had to be established experimentally.

The sampling constant exposures were repeated at 9 ±1°C, 25 ±1°C, and 48 ±2°C and the samplers desorbed and analyzed by GC/MS and GC/ECD. The low temperature was achieved by surrounding the exposure chamber with a styrofoam enclosure filled with ice, which was drilled with holes that allowed access to measuring probe holes. The inner chamber temperature was continuously measured with a calibrated thermometer and the RH with a calibrated hygrometer during the exposures. The high temperature was attained by placing the exposure chamber into a Thermolyne Series 5000 Incubator after taking off its glass door and replacing it with a same-dimensioned Plexiglass door (thickness 1 mm) from a print frame that was drilled with holes to allow access of the measuring probes and power wires. The mass collected as the O-oxime was plotted against minutes × milligrams per milliliter. The efficiency relative to that expected for the O-oxime equivalent from using Equation 2 for aldehyde was then calculated for each temperature.

Storage Stability

Vapor spiked samplers exposed equivalently at the TLV × 8 hr were stored using the 3M sampler directions[32,33] at 4.0 ±0.5°C and 25 ±1°C for 3 months with a sampler analyzed every week by GC/MS and GC/ECD.

Shelf Life

Samplers were subjected to 1 hour of exposure at the TLV 8-hr equivalent for a week for 3 months to compare the amounts desorbed relative to when the coated sorbent was fresh.

Statistics

All internal comparisons in each section of the protocol were subjected to analysis of variance Type I and II procedures to detect significant differences at $\alpha = 0.05$ and significant interactions.[40] The p values for intercept significance for linear regressions relative to (0,0) were performed using the Student t distribution assuming the same standard deviation as the experimental result for zero.[41]

RESULTS

Table II gives the theoretical sampling constants at the three different temperatures investigated for both valeraldehyde and acrolein, based on the theoretical diffusion coefficients calculated with Equation 2. For valeraldehyde, the diffusion coefficient at 25°C and 1 atm was calculated as follows:

$$D_{\text{air-valeraldehyde}} = 0.001 \times 298^{1.75} \times (1/28.8 + 1/86.13)^{0.5}/[1 \times (19.7^{1/3} + 108.71^{1/3})^2] = 0.083 \text{ (cm}^2\text{/sec)}$$

For acrolein, the diffusion coefficient at 25°C and 1 atm was calculated as follows:

$$D_{\text{air-acrolein}} = 0.001 \times 298^{1.75} \times (1/28.8 + 1/56.06)^{0.5}/[1 \times (19.7^{1/3} + 67.05^{1/3})^2] = 0.108 \text{ (cm}^2\text{/sec)}$$

The results for both valeraldehyde and acrolein are summarized in Table III for the sampler covered with the silicone membrane. The desorption efficiency (n=6) for spiked PFBHA O-oxime was about 95.9 ±5.4%, whereas the desorption efficiency for wet aldehyde spiking under a number of conditions (n=6, each) was (94.0 ±3.6%, not significantly different at $\alpha = 0.05$ (p=0.6388).

Passive samplers with and without diffusive membranes were tested against valeraldehyde vapor under the same exposure conditions with respect to face velocity to assess the effect of the membrane on the sampling efficiency relative to that expected

TABLE III. Results for Valeraldehyde and Acrolein for the Validation Protocol of Table I

Parameter	Valeraldehyde	Acrolein
O-oxime desorption efficiency	95.6 ± 5.4%	107.2 ± 8.1%
Wet aldehyde desorption efficiency at 25°C at TLV × 8 hr		
At 25 ± 1°C for 5 days	91 ± 15%	103.1 ± 7.2%
4.0 ± 0.5°C for 2 days then 25 ± 1°C	93.7 ± 2.8%	96.1 ± 3.1%
4.0 ± 0.5°C for 5 days	98.5 ± 6.7%	99.4 ± 8.2%
4.0 ± 0.5°C for 5 days then desorption at 4°C	92 ± 10%	95.2 ± 4.1%
Average	94.0 ± 3.6%	98.5 ± 7.3%
Critical face velocity	15–20 ft/min (7.6–10.2 cm/sec)	15–20 ft/min (7.6–10.2 cm/sec)
Capacity (μmoles aldehyde equivalent)		
Wet	28.0 ± 4.8	25.9 ± 5.4
Dry	32.9 ± 3.5	29.1 ± 2.8
Intermittent exposures tests[A] (at 3 ± 1% RH and 25 ± 1°C)		
Generation: 0–1 hr; no generation 1–5 hr	72.7 ± 6.7%	97.5 ± 6.6%
No generation: 0–3.5 hr; generation 3.5–5 hr	77.9 ± 5.3%	96 ± 12%
Generation: 0–1, 1.5–2.8, 3.8–5 hr with no vapor generation in between	73.0 ± 2.4%	95.1 ± 8.6%
	78.1 ± 1.2%	94 ± 10%
Generation: 0–1 hr TLV continuous for 8 hr	75.4 ± 3.9%	94.0 ± 8.8%
Relative humidity tests[A] (at 25 ± 1°C)		
3 ± 1% RH	73.8 ± 3.2%	101.2 ± 6.6%
36 ± 2% RH	75.2 ± 2.5%	Not done
79 ± 2% RH	74.5 ± 6.8%	91.5 ± 12[B]
Temperature tests[A] (at 3 ± 1% RH)		
9 ± 1°C	74.1 ± 1.0%	99.7 ± 8.1%
25 ± 1°C	73.8 ± 3.2%	102.2 ± 6.6%
48 ± 2°C	76.7 ± 1.7%	96.3 ± 1.2%
Overall average	74.4 ± 7.2%	99.4 ± 7.2%
Overall experimental sampling constant (mL/min) 9–48°C and 3–79% RH	4.43 ± 0.19	7.73 ± 0.57
Collected sample stability over 6 months (% of original at start for n = 3)		
25 ± 1°C	95.7 ± 6.2	101.8 ± 1.9[C]
4.0 ± 0.5°C	91.9 ± 5.9	95.2 ± 6.6[C]
Sampler shelf life (% response relative to exposure when sampler was just made for n = 3)		
25 ± 1°C over 3 months	92.4 ± 8.1	93.5 ± 9.9

[A] Sampling efficiency (%) relative to expected from Equations 1 and 2 at TLV × 8 hr equivalent exposures, and n = 6 for each condition.
[B] At 90% RH.
[C] At 4 weeks.

from Equations 1 and 2. Both passive samplers with (n=21) and without (n=15) diffusive membranes showed increasing mass collected as face velocity increased (Figure 3). If the data for face velocity below 20 ft/min with and without membrane were excluded, the passive samplers without diffusive membranes still had a trend of increasing sampling efficiency as the face velocity increased. However, the sampling efficiency for the sampler with membrane was independent of face velocity. Figure 3 also shows that the critical face velocity for valeraldehyde (n=21) and acrolein (n=26) is between 15–20 ft/min (7.6–10.0 cm/sec). Even at zero face velocity, the efficiency for valeraldehyde was still 52–57% of that calculated from Equations 1 and 2 relative to 70–80% above the critical face velocity (mean 76.3 ±4.6%). Similarly, the efficiency for acrolein at zero face velocity was 25–35% compared with 90–110% above the critical face velocity. These results confirm that theory does not necessarily predict sampling constants, which must be measured directly.

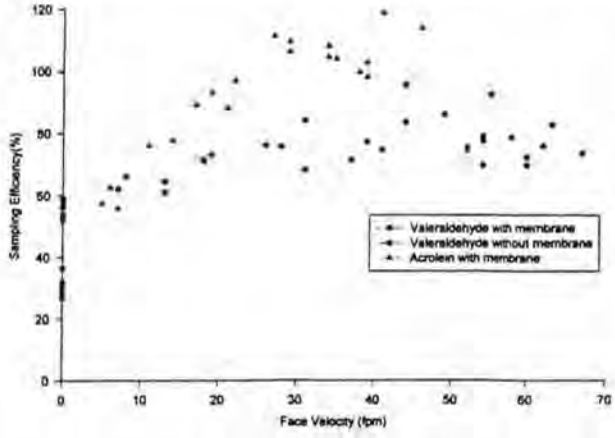

FIGURE 3. Face velocity behavior for acrolein and valeraldehyde

Figure 4 shows the saturation profile of the collection element

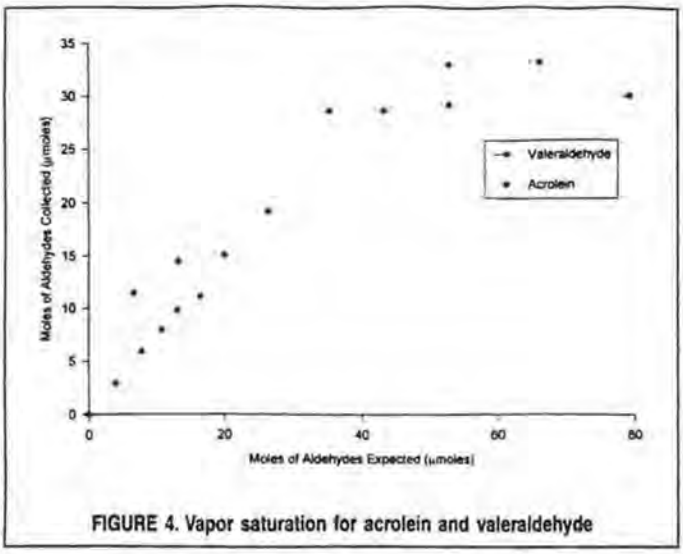

FIGURE 4. Vapor saturation for acrolein and valeraldehyde

in terms of moles of aldehyde vapor collected versus aldehydes expected from Equations 1 and 2. From Figure 4 and Table III, the vapor capacity is about 33 μmoles of aldehyde (n=40 and n=24 for valeraldehyde and acrolein, respectively). For valeraldehyde this is equivalent to a 50-ppm exposure for 60 hours or a capacity about eight times the ppm-hours equivalent to 8 × TLV-TWA. The capacity on wet spiking of aldehyde is about the same. The same capacity was shown for the sensing element with a membrane or within its 3M plastic holder. The same results were obtained for acrolein. Thus, the total number of moles of exposure to aldehyde that the sampler can accommodate must not approach 30 μmoles of aldehydes with one carbonyl group.

Figure 5 shows the direct reading organic vapor analyzer time profiles for the intermittent vapor exposures for valeraldehyde. The half-times to maximum concentration varied between 5–10 min and the half-times to decrease to baseline varied between 10–15 min. Since the ppm-hour were calculated from taking the area under the ppm versus time curve rather than assuming exposure time matched generation time, the actual exposure dose was calculated rather than the theoretical one. This was impossible for acrolein because of the inadequate sensitivity of the organic vapor analyzer, and that situation necessitated a dynamic sampling method that automatically integrated over the exposure periods (n=3, each condition). The acrolein sampling rate measured for the passive sampler was determined from the known flow rate of the dynamic sampler for each exposure protocol. The measured sampling

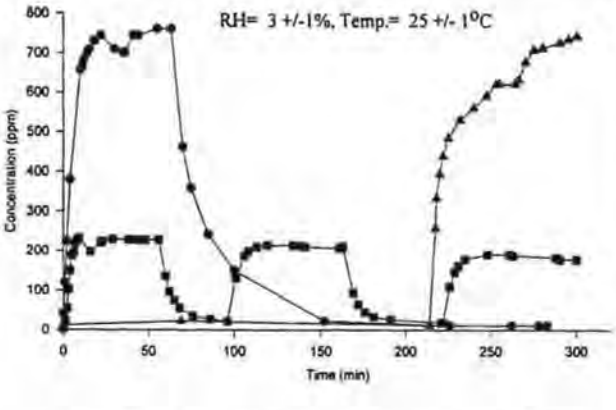

FIGURE 5. Intermittent exposures for valeraldehyde

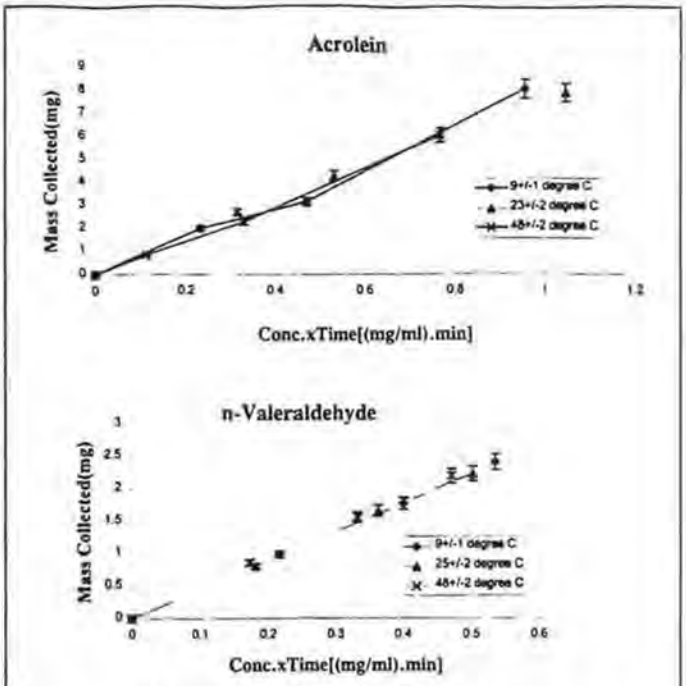

FIGURE 6. Temperature dependence of passive sampling constant for acrolein and valeraldehyde from 9 to 48°C

rates for the passive sampler did not differ statistically from the predicted ones from Equations 1 and 2. As shown in Table III, all of the sampling efficiencies relative to what was expected from Equations 1 and 2 for these three types of exposures for each compound did not differ significantly, and also did not differ from those determined for 1 hour exposure at the TLV × 8-hr equivalent, or for the TLV over 8 hr (n=6 for each condition). Thus, 1-hour exposures at the TLV × 8-hr equivalent could be done instead of 8-hr exposures at the TLV for each compound, saving much time, and allowing use of direct reading instruments at the high concentration end to validate the generation technique.

There were no effects of temperature (Figure 6 and Table III) on mass collected of aldehyde equivalent (n=6 for each temperature). Table III also shows that RH did not cause different sampler efficiencies at 25°C (n=6 for each RH). There were no effects of shelf life or storage at either 4 or 25°C (Table III). Thus, the pellet does not need refrigeration before or after exposure. However, it still needs rewrapping in aluminum foil and/or being placed in a sealed container to prevent adventitious sampling.

The masses collected of aldehyde equivalent m_i for aldehyde i (in mg) at exposure concentration c_i (in mg/mL) over exposure time t (in min) were not significantly different at $\alpha=0.05$ for all intermittent and continuous exposures of the same ppm-hours for the sampler with its membrane and within its 3M plastic holder (Table III). Temperatures and RH also had no significant effects (Table III). Therefore, pooling all the homogeneous data for the same ppm-hours for all conditions gives Equations 6 and 8 for valeraldehyde v and acrolein a, respectively, as regression equations.

For valeraldehyde, n=60,

$$m_v = (4.43 \pm 0.19)\, c_i t + (0.003 \pm 0.021) \quad (6)$$

or, since the intercept is not significantly different from zero at $\alpha=0.05$ (p=0.54),

$$c_v = m_v/t(4.43 \pm 0.19) \quad (7)$$

The coefficient of variation (CV) for c_v is therefore 4.3%. For acrolein, n=54,

$$m_a = (7.73 \pm 0.57)\, c_a t + (-0.007 \pm 0.071) \quad (8)$$

or, since the intercept is not significantly different from zero at $\alpha=0.05$ ($p=0.81$),

$$c_a = m_a/t(7.73 + /-0.57) \quad (9)$$

The CV for c_a is therefore 7.4%.

DISCUSSION

The most important results were the nondependence of sampling efficiency relative to continuous 8-hr sampling for intermittent exposures, RH, storage, or shelf life. This is consistent with the behavior of the dynamic sampler.[19] The critical face velocity for most chemisorptive passive samplers for formaldehyde is <15 ft/min (<10 cm/sec).[25,29] However, storage stability is a problem for these other passive samplers[26] in contrast to the new one developed in the present study. In a comprehensive comparison of formaldehyde samplers, only the GMD 570 sampler housed in a ProTek-like organic vapor sampler badge based on DNPH passed all tests at 0.3, 0.5, and 1.0 ppm for 8 hr[26] in a protocol that also examined other passive samplers like those from Pro-Tek Series II Type C-60, Crystal Diagnostics Airscan®, 3M 3721 monitors, Assay Technology ChemChip® and Sensidyne Gastec 91D. Variable biases were observed even for the dynamic methods like Occupational Safety and Health Administration Method 52 (DNPH coated silica gel available now from Supelco as Orbo 24 tubes) and DNPH coated reverse phase C18 tubes (now commercially available from the Waters Chromatography Division of Millipore Corp.) relative to the NIOSH 3500 dynamic method, and a gravimetric permeation tube source. All the DNPH-based devices except the GMD badges and including the dynamic sampling tubes read low at 27% RH under the same formaldehyde TLV-exposure time conditions relative to 75% RH. These investigators did not generate RH atmospheres around 3% as was done in the present study. The reason for the success of the GMD badge is probably because its method of analysis corrected for an internal blank that may also be RH sensitive, and the use of phosphoric acid and glycerin in the coating process. Though the critical face velocity was not determined, the utilized velocity of 51 ft/min to expose passive samplers would be well above the critical face velocity observed by other workers. No temperature dependence nor shelf-life experiments were done, however.

The inadequacy of Equation 2 to predict temperature effects for the PFBHA-coated sorbents (Table II) was surprising. Since the liquid aldehyde and O-oxime desorption efficiencies did not differ and were quantitative, factors affecting the reaction other than those involving aldehyde diffusion must be dominant. The solid phase/gas phase chemical reaction is most probably influenced by the large excess of PFBHA, by the aldehydes having affinity for the uncoated solid sorbent,[36] and the probable presence of heterogeneous catalysis[38] thus making the activation energy low in Equation 5 and hence not dependent on temperature. Such a characteristic for a passive sampler is very desirable. Initially it was thought that the desorption step at 25°C could account for these results, but desorption at 4°C for wet aldehyde spiking was still not significantly different from desorption at 25°C. In contrast, the direct reading passive indicator from AirChem Technologies Monitoring Card System (Environetrics Products Co.) showed uncontrolled biases at 16 and 35°C.[29]

The sampling efficiency of valeraldehyde was about 75% of the theoretical value calculated from Equations 1 and 2, even though that number had a small CV of 9.7%. The reaction efficiency for valeraldehyde wet spiking of the dynamic sampler was between 95 to 102%, compared with 91 to 99% for the passive sampler, both not being different statistically. The absolute recovery for valeraldehyde vapor in the dynamic method varied with flow rate, 10 mL/min being far more efficient (efficiency of about 100%) than 50 mL/min (efficiency 71 to 85%). This suggested that contact and reaction times were important as well as the degree of packing of the coated sorbent bed with the possibility of surface effects. The packing factor is uniform for the passive sampler. Therefore, surface reaction factors have to be responsible since the desorption of oxime after spiking liquid aldehyde at 4°C had the same efficiency as at 40°C.

The major alternative passive air sampler based on DNPH has been validated for formaldehyde only, but not other aldehydes.[26-29] The DNPH method requires HPLC for analysis, which does not distinguish alpha-hydroxycarbonyls and the corresponding dicarbonyls (for example, glycolaldehyde/glyoxal and hydroxyacetone/methylglyoxal), and similar carbonyls (for example, acrolein, acetone, and propanal). In addition, the absorption spectra vary with pH since stopped-flow HPLC spectra often differ from those of acidified or basic standards, the absorption spectra also vary with the amount of DPNH present, and there is a pronounced temperature effect in solution.[42] The dynamic DPNH silica gel air sampling method has been used extensively,[13,27,43-45] but little attention has been paid to the factors that affect HPLC hydrazone analysis until recently.[42] Ozone is also known to interfere with the DNPH coated silica gel air sampler reaction with air formaldehyde.[45] In contrast, greater sensitivity and resolution are possible for GC/MS and GC/ECD using PFBHA O-oxime derivatives, thus allowing unknowns to be identified more easily than through the less sensitive liquid chromatography/mass spectrometry method.[31] In addition, reaction efficiencies decrease for dry air relative to humid air for the dynamic DNPH sampling method using coated C18 reverse phase cartridges.[27] The latter effect is not shown by Tenax GC or Tenax TA coated solid sorbents used in the dynamic[19] or passive sampling modes, nor by the coated DNPH glass fiber filter passive monitor (uptake rate 60 mL/min) impregnated also with phosphoric acid and glycerin.[26] The related GMD sampler (uptake rate 25 mL/min) has not been investigated for its temperature dependence.

The current status of the NIOSH diffusive sampler evaluation protocol[22] has recently been reviewed.[46] One of the review's refrains is that the NIOSH protocol is extremely cumbersome, but a bilevel (rather than a factorial) design suffices as a screening tool if physical adsorption is the basis of the sample collection and retention.[47] Physical adsorption shows a low temperature coefficient of the diffusion coefficient of about 2.5% from 25 to 10°C or from 25 to 40°C, both well within experimental errors[46] and similar to the results of the present study relative to temperature. The present study also shows that a still leaner protocol will suffice if reverse diffusion is not important, but applies only for strong chemisorptive processes. The present validation technique depends on the selection of the extremes to be encountered, here the lowest and highest TLV-TWA of the aldehydes of hygienic interest assuming that the chemisorptive efficiency is about the same, which it appeared to be from the dynamic sampler study[19] and

the aqueous solution investigation.[35] Experimental sampling rates also still have to be determined since theoretical results using Equations 1 and 2 were not completely predictive (even though the discrepancy for valeraldehyde was within the NIOSH acceptability criterion), a finding also concluded by other investigators.[46] The chemisorptive nature of the sampler, with its nonvolatile reaction product, makes moot any argument regarding whether the sampler has to be capped to account for intermittent exposures as occurs for samplers operating solely by physical adsorption.[48]

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AMERICAN INDUSTRIAL HYGIENE ASSOCIATION JOURNAL 60:474–479 (1999) Ms. #862

Effect of Respirator Inspiratory Resistance Level on Constant Load Treadmill Work Performance

AUTHORS
Arthur T. Johnson
William H. Scott
Christopher G. Lausted
M. Benhur Benjamin
Karen M. Coyne
Manjit S. Sahota
Monique M. Johnson

Biological Resources Engineering, University of Maryland, College Park, MD 20742-5711

Respirator inspiratory resistance can affect performance times, especially when the experiment is optimized to elicit respiratory stress. Twelve subjects performed on a treadmill at constant speeds and grades chosen to result in performance times of 5–15 min. Six levels of inspiratory resistance were used, ranging from 0.78 to 7.64 cm H_2O•sec/L. The results showed that performance times decrease linearly with resistance level, and no threshold resistance value is apparent. Inspiratory resistance also induces hypoventilation, with lower minute volumes and lower oxygen consumption values at higher resistances. These trends are also linear. From these results, there is no value for inspiratory resistance that can be given as a design goal. Other parameters such as weight and space may dictate filter resistance values, and these, in turn, will lead to determined performance degradations.

Keywords: resistance, respirator, work

The effects of respirator mask inhalation resistances on performance of wearers has long been of interest. If the performance-resistance relationship could be established, then many questions about respirator filter design could be answered. Unfortunately, past experimental data are flawed for one or more reasons. Of special importance is planning the experiments to be most sensitive to respiratory stress.

Johnson and Cummings[1] were the first to suggest that performance times in the range of 5–15 min would involve a work rate that would be most sensitive to respiratory stress. Work rates more severe than that would be expected to be most sensitive to cardiovascular loading, and less severe work rates would be most sensitive to thermal stress. Stemler and Craig[2] performed a series of experiments wherein they tested both performance time and exhalation time hypotheses using a set of six respirator conditions. Their inspiratory resistance levels ranged from 0 to 3.7 cm H_2O•sec/L and they obtained treadmill exercise performance ratings (performance time with respirator divided by the time without a respirator[3]) ranging from 100 to 62 (meaning that, for a resistance of 3.7 cm H_2O•sec/L, a performance time of 62% of the control condition, bareheaded, would be expected). They concluded that the general range of exercise time given by Johnson and Cummings seems to elicit results very sensitive to respiratory resistance levels.

Caretti and Whitley[4] performed a test of treadmill performance time at 80–85% of maximal aerobic capacity for a control condition and four different inspiratory resistances ranging from 1.4 to 3.5 cm H_2O•sec/L. They did not use a full facepiece respirator, but applied their resistances to a half-mask used for exercise studies. Treadmill speed and grade remained constant throughout each test session, but different speeds and grades were used for each resistance condition to elicit the required 80–85% of $\dot{V}O_2$ max. Their performance times ranged down to 30% of the control condition for their highest resistance.

Others[5,6] who intended to test the effects of inspiratory resistance on exercise performance time have not recognized the need to load subjects at a sufficiently high work rate to elicit maximum response. They thus did not detect large differences in performance times for substantial ranges of inspiratory resistance. When, based on these results, they concluded that inspiratory resistance had little effect on performance while wearing respirators, they were both right and wrong: right, because unless workers are required to work at rates approaching 80–85% of

APPENDIX 2

Applied Occupational and Environmental Hygiene
Volume 14: 255–262, 1999

1047-322X/99 $12.00 + .00

A New Passive Sampler for Regulated Workplace Aldehydes

Shih-Wei Tsai and Shane S. Que Hee

Department of Environmental Health Sciences and Center for Occupational and Environmental Health, School of Public Health, University of California at Los Angeles, Los Angeles, California

A new solid sorbent passive air sampler for aldehydes had a silicone membrane atop a cylindrical diffusion path of 1.1 cm length and 1.3 cm diameter above a 10 percent (w/w) O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) Tenax TA pellet. Known vapor concentrations of Occupational Safety and Health Administration (OSHA)-regulated aldehydes near their permissible exposure limits were generated from a syringe pump dynamic air dilution system that was connected to an exposure chamber. The O-oxime derivatives from aldehyde reaction with PFBHA were desorbed with hexane, and quantified by capillary gas chromatography/mass spectrometry (GC/MS) or gas chromatography/electron capture detection (GC/ECD). The capacity for aldehydes with one carbonyl group was 30–35 μmoles, and 15 μmoles for the dialdehyde, glutaraldehyde. The experimental sampling rates in mL/min were 8.86 ± 0.38, acetaldehyde; 11.69 ± 0.32, chloroacetaldehyde; 7.85 ± 0.19, crotonaldehyde; 9.97 ± 0.10, formaldehyde; 6.47 ± 0.42, furfural; and 4.46 ± 0.15, glutaraldehyde. Other data on valeraldehyde and acrolein have shown that the sampling constants were independent of face velocity between 0.1 to 0.35 m/s (20 to 70 fpm), temperatures between 9 to 48°C, RH between 3 to 79 percent, and intermittent sampling exposure pattern.

Keywords Adsorption, Aldehyde, Aldehyde Oxime, Gas Chromatography, Passive Sampler, Personal Sampling

INTRODUCTION

Aldehydes (R-CHO where R is alkyl, aromatic, or alicyclic) are widely used industrial chemicals that are also detected in the environment as products of combustion,[1–6] water disinfection,[7] and biological oxidation.[8] Aldehydes irritate mucous membranes.[9] Formaldehyde, acetaldehyde, furfural, and crotonaldehyde are animal carcinogens.[10] Adverse exposure effects of formaldehyde and glutaraldehyde are documented to embalmers,[11,12] operating theater personnel,[13] and pathologists.[9] The exposure and the health effects of formaldehyde have been reviewed elsewhere.[14,15]

Aldehyde vapors are usually sampled with solid sorbents.[2,16–19] The National Institute for Occupational Safety and Health (NIOSH) method uses XAD-2 resin coated with 2-(hydroxymethyl)piperidine. Nonreactive C_3-C_5 aldehydes are not collected quantitatively,[16] and volatile acids reduce loading capacity. The 2,4-dinitrophenylhydrazine (DNPH) method is used by the Environmental Protection Agency (EPA)[18] and the American Society for Testing and Materials (ASTM).[20] The DNPH methods potentially allow specific quantitation of different aldehydes and ketones through high performance liquid chromatography (HPLC)/ultraviolet (UV) detection of their hydrazones. DNPH does not react quantitatively with conjugated aliphatic aldehydes. The hydrazones are light-sensitive, and their recoveries are variable after liquid aldehyde spiking.[2] Some passive samplers have been developed for the lower molecular weight aldehydes and ketones based on liquid systems,[22–24] and on solid sorbents coated with DNPH.[22,25–28]

O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) has been used to react quickly and quantitatively with aldehydes in water to form O-oximes suitable for picogram (pg) detection by gas chromatography/mass spectrometry (GC/MS) and gas chromatography/electron capture detection (GC/ECD).[21] The PFBHA method also has been used to chemisorb aldehydes by dynamic air sampling.[19] We have previously reported[29] on a new passive sampler design for *n*-valeraldehyde and acrolein based on the PFBHA method. We now extend the method to other aldehydes regulated by the Occupational Safety and Health Administration (OSHA).

EXPERIMENTAL METHODS

Materials

The aldehydes investigated were acetaldehyde (99.5 percent w/w), chloroacetaldehyde (50 percent in water), crotonaldehyde (98 percent), formaldehyde (37 percent in water), furfural

(98 percent), and glutaraldehyde (50 percent in water), all from Aldrich, Milwaukee as was the internal standard decachlorobiphenyl. Optima hexane, Optima methanol, nitric acid, activated charcoal, molecular sieves, and Drierite were from Fisher Scientific, Tustin, Los Angeles. PFBHA (Lancaster Laboratories Inc., Lancaster, Pennsylvania), Tenax TA 80/100 mesh (Alltech Associates, Deerfield, Illinois), and 5 percent (v/v) methane/argon (Alphagaz, Los Angeles, California) were also used.

Equipment

Lengths of Pyrex tubing (7 cm × 5-mm ID), Pyrex® glass wool (Soxhlett-extracted in methanol and then hexane), 4-mL Kimble vials (PTFE-lined screw caps), 10-μL Hamilton syringes, gas-tight Hamilton syringes, a Parr 2811 bench manual pellet press, 3M Model 3500 OVM passive sampler, and a Model 11 Harvard syringe pump were from Fisher Scientific. Dupont P30A personal sampling pumps, Tedlar gas bags (SKC Inc., Eighty Four, Pennsylvania), and a Whatman Zero Air generator (Balston Inc., Haverhill, Massachusetts) were also used.

A Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, California) equipped with a 30-m × 0.32-mm ID 1-μm film DB-1701 chemically bonded fused-silica capillary column was linked to either a Hewlett-Packard 5988A mass spectrometer or a ^{63}Ni-electron capture detector (ECD). The temperature for the injector, ECD, and GC/MS transfer line was 250°C. The MS ion source temperature was 260°C. The column temperature program for glutaraldehyde was: solvent delay, 5 min at 105°C; 105°C for 0.5 min, 105°C to 220°C at 70°C/min, and holding then for 40 min. The temperature program for other aldehydes was: solvent delay 5 min at 105°C, 105°C for 0.5 min, 105°C to 220°C at 10°C/min, and holding then for 10 min. Selective ion monitoring (SIM) used m/z 181 and total ion monitoring m/z 50-500. Quantitation used both E- and Z-isomer areas. The carrier gas for GC/MS was helium and for GC/ECD 5% methane/argon at 3.0 ± 0.4 mL/min. The ECD signal was displayed with a Hewlett-Packard 3396 integrator.

Methods

A 13-mm-diameter and 0.2-cm-thick pellet of PFBHA-coated Tenax TA (10 percent, w/w) was made by the hand press. The sampler had a silicone membrane and a diffusion path length of 1.1 cm (Figure 1). Figure 2 presents the vapor generator, air dilution system, and exposure chamber.[29] The air generator was connected to the vapor and water generation syringe pumps set at known plunger speeds. Heating tape wrapped around the outside of the stainless steel tubing at the needle exit from the syringe pumps ensured total volatilization of organic vapor or water. The two streams were then routed through a stainless steel T-joint adapter connected by Teflon® tubing to a Greenburg-Smith impinger mixing chamber. Teflon tubing then conveyed the diluted mixed organic vapor into the exposure chamber through a

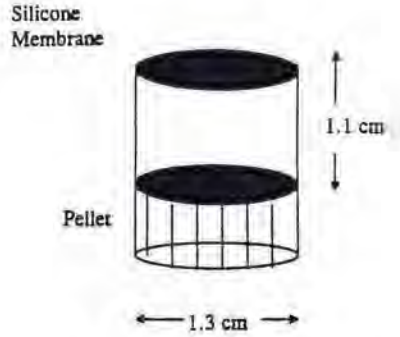

Diagram of Sampler

FIGURE 1
Cross-section of the passive sampler.

leakless hole bored on the side wall near the chamber bottom to just underneath the chamber fan blades under the ceramic metal plate. Six samplers were placed horizontally on the plate, each with a nearby closable hole in the chamber wall for insertion of calibrated probes to measure RH, temperature, organic vapor concentration, and face velocity. For acetaldehyde of boiling point 21°C,[30] the syringe pump was surrounded by ice cubes and blue ice at 4 ± 1°C to stop acetaldehyde liquid evaporation before entering the dilution system.

Aqueous aldehydes, including formaldehyde, glutaraldehyde, and chloroacetaldehyde are easily polymerized. Therefore, exact purity was determined for generation purposes using the NIOSH back-titration method with 1.13 M sodium sulfite.[16]

Synthesis of PFBHA O-Oximes

The PFBHA O-oximes were synthesized by methods detailed elsewhere[19] except for glutaraldehyde. A molar ratio of 8:1 of PFBHA (90.5 mg; 0.36 mmol) to glutaraldehyde (50 percent solution; density 1.007 ± 0.026 g/ml) was used to synthesize the di-substituted glutaraldehyde PFBHA O-oxime. Attempts were made to make the monosubstituted O-oxime by varying the ratio of aldehyde to PFBHA and reaction times. White precipitates formed immediately in all cases. The mixtures were centrifuged for 5 minutes. Subsequent steps were the same as for other O-oximes.[19]

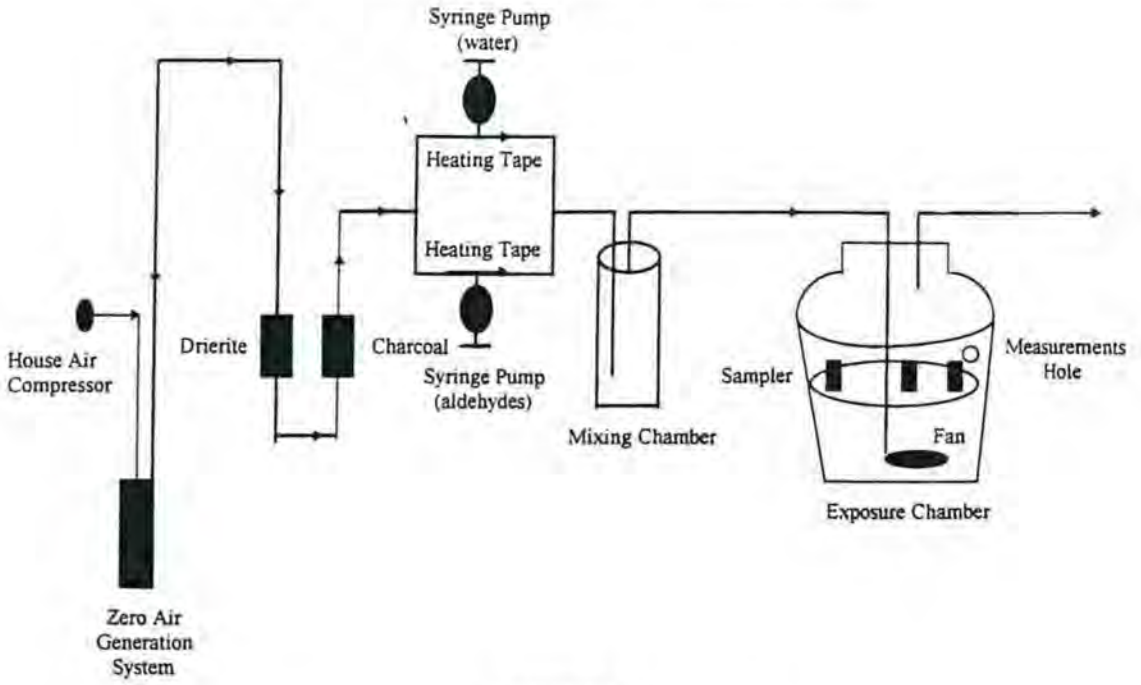

FIGURE 2
Aldehyde vapor generation and exposure chamber system.

Aldehyde Diffusion Coefficients and Sampling Constants

The dependence of the diffusion constant on molecular weight and temperature is expressed through equation (1):[31]

$$D_{AB} = \frac{0.00143 \times T^{1.75}}{PM_{AB}^{1/2}\left[\left(\sum_V\right)_A^{1/3} + \left(\sum_v\right)_B^{1/3}\right]^2} \qquad [1]$$

where D_{AB} is the binary diffusion coefficient of analyte in air in cm^2/s at T; T is temperature, K; M_A and M_B are molecular weight, g/mol; $M_{AB} = 2[(1/M_A) + (1/M_B)]^{-1}$; P is the external pressure, bar; $\sum_V$ is the summation of atomic diffusion volumes, unitless; i is all the contributing species; A is air; B is the analyte; $(\sum_V)_A$ is 19.7.[31] For acetaldehyde H_4C_2O, the $\sum_V$ is:[31]

$$\left(\sum_V\right)_{H_4C_2O} = 4 \times 2.31^H + 2 \times 15.9^C + 1 \times 6.11^O = 47.2 \qquad [2]$$

where[31] H = 2.31 is the atomic diffusion volume increment for hydrogen; C = 15.9 is the atomic diffusion volume increment for carbon; O = 6.11 is the atomic diffusion volume increment for oxygen.

The method does not distinguish between keto and enol isomers. The diffusion coefficient of acetaldehyde at 25°C and 1 atm (1.01 bar) is:

$$D_{Air-Acetaldehyde} = \frac{0.00143 \times 298^{1.75}}{1.01 \times [2 \times (1/29.0 + 1/44.1)^{-1}]^{1/2} \times [19.7^{1/3} + 47.2^{1/3}]^2} = \frac{30.56}{1.01 \times 5.92 \times 39.8} = \frac{30.56}{238.02} = 0.128 \text{ (cm}^2\text{/sec)} \qquad [3]$$

The theoretical sampling constant k is implicated in Fick's first law of diffusion as shown in equation (4) in its form for a cylindrical open tube:[32]

$$dm/dt = (D_{AB}\ A/L)(c_{air} - c_{surf}) = k(c_{air} - c_{surf}) \qquad [4]$$

where dm/dt is the steady state mass sampling rate or mass transfer rate, weight/time; A is the effective cross-sectional area of the sampling element, cm^2; L is the effective path length where diffusion control prevails to the sampling element from the exposing atmosphere, cm; c_{air} is the air concentration of the analyte, weight/cm^3; c_{surf} is the air concentration of analyte just above the sampling surface in the same units as c_{air}; k is the sampling constant of the analyte equal to $(D_{AB}\ A/L)$, cm^3/time.

For the present sampler, A/L is $(\frac{1.3}{2})^2 \times \pi \times \frac{1}{1.1} = 1.2$ cm.

Reaction Efficiency/O-Oxime Recovery for Wet Spiking of Aldehyde

Liquid aldehydes equivalent to two times the PEL-8 hour mass as determined from the theoretical sampling constant (or

two times the STEL-15 min) were spiked on methanol on to the pellets. The spiked pellet was held overnight in a desiccator containing Drierite to allow the methanol to evaporate before desorption with 2.0 mL hexane at room temperature over 2 hours with 30 seconds of ultrasonication at every half-hour before analysis by GC/MS or GC/ECD using synthesized aldehyde oximes of known purity.

Face Velocity, Relative Humidity, Temperature, and Capacity

For all experiments, the face velocities were above 0.10 m/s (20 fpm), the critical face velocity of the sampler[29] while the range of face velocities encountered in a typical workplace is above 0.10 m/s.[22] The relative humidity (RH) was $3 \pm 1\%$, that is, no humidification was done. The temperature was $22 \pm 1°C$. All data were corrected to 25°C and 1 atmosphere pressure. All saturation capacities were determined through liquid aldehyde spiking in methanol, the O-oxime desorbed by hexane, and quantification by GC/MS or GC/ECD. The vapor capacities were not done here since liquid spiking and vapor spiking had the same aldehyde saturation capacity.[29]

Vapor Exposures

The ppm-hour levels of exposure were equivalent to 0, 0.5, 1 and 2 times the PEL for 8 hours, or 15 min for the STEL. Dynamic sampling at 50 mL/min flow rate was utilized as a reference method after its sampling efficiency was evaluated for each aldehyde using the static gas bag method.[19] The O-oxime was desorbed with hexane, and quantified by GC/MS or GC/ECD. The moles desorbed was plotted against (μmole/mL) × min.

Statistics

All internal comparisons were subjected to analysis of variance (ANOVA) types I and II to detect significant differences at $p < 0.05$ and significant interactions.[33]

RESULTS

Table I shows the results of O-oxime syntheses in terms of GC/MS purities, raw reaction yields, and yields corrected for purities. GC/MS purities correct for the presence of pentafluorobenzaldehyde, pentafluorobenzyl alcohol, and excess PFBHA in the O-oximes.[34] All purities exceeded 92 percent. The corrected reaction yields are based on the assumed 1:1 stoichiometry except for glutaraldehyde (1:2). All yields exceeded 81 percent. Average percent purities/reaction yields were $95.8 \pm 2.5/91.1 \pm 2.3$, respectively. The E- and Z-isomers had the same molecular ion and fragmentation pattern, though different retention times.[34] No monoaldehyde derivative was isolatable for glutaraldehyde even when aldehyde was rate limiting.

Table II shows the results of reaction efficiency/O-oxime recovery for wet spiking of aldehydes, quantifying by pure O-oxime standards. The results for acrolein and valeraldehyde are also included for comparison.[19] Desorption efficiencies for spiked O-oximes were not done because there was no significant difference between O-oxime spiking desorption efficiencies and wet aldehyde spiking recoveries from previous work.[29] The results of dynamic sampling for known gas bag vapor concentrations are also shown in Table II relative to pure O-oxime standards. Although the wet spiking recoveries always exceeded 89 percent for all compounds (mean ± SD: 99.8 ± 6.5 percent), only the monoaldehydes had vapor sampling efficiencies > 85 percent.

TABLE I
GC/MS purity and yield results for aldehyde O-oxime syntheses

	GC/MS purity (%)	Raw yield (%)	Yield corrected for purity (%)
Acetaldehyde	99.5 ± 2.5	90.0 ± 2.0	89.6 ± 2.3
Chloroacetaldehyde	96.0 ± 1.2	91.7 ± 1.2	88.0 ± 1.1
Crotonaldehyde	96.6 ± 2.2	91.2 ± 2.6	88.1 ± 2.0
Formaldehyde	93.2 ± 1.4	90.5 ± 2.3	84.3 ± 1.3
Furfural	97.0 ± 1.2	95.0 ± 2.1	92.2 ± 1.1
Glutaraldehyde	92.7 ± 3.2^a	88.0 ± 2.5^a	81.6 ± 2.8
Average	95.8 ± 2.5	91.1 ± 2.3	87.3 ± 3.8

[a] Di-substituted glutaraldehyde-PFBHA oxime.

Figures 3 and 4 show the saturation profiles of the collection element in terms of moles of aldehyde collected backcalculated from moles of O-oximes quantified versus moles of aldehydes expected (equation [4]). As for valeraldehyde and acrolein,[29] the capacities for monoaldehydes were 30–40 μmol. Glutaraldehyde capacity was 10–15 μmol. Thus, the moles of exposure to aldehyde that the sampler accommodated must not approach 30 μmol for $30 \times 10^{-6} \times 6.0234 \times 10^{23} = 1.8 \times 10^{19}$ total carbonyl groups.

TABLE II
Efficiencies from wet spiking and dynamic sampling in terms of O-oxime recovered relative to pure O-oxime standards

	Wet spiking (%)	Dynamic sampling (%)
Acetaldehyde	96.3 ± 3.2	93.0 ± 3.6
Acrolein[a]	99.0 ± 6.0	86.0 ± 5.0
Chloroacetaldehyde	112.7 ± 4.6	114.6 ± 2.5
Crotonaldehyde	101.3 ± 7.9	115.8 ± 3.6
Formaldehyde	99.9 ± 9.8	107.5 ± 4.6
Furfural	89.3 ± 3.4	94.4 ± 7.9
Glutaraldehyde	100.9 ± 1.7	23.3 ± 2.6
Valeraldehyde[a]	99.0 ± 3.0	85.0 ± 2.0
Average ± SD	99.8 ± 6.5^b	100.0 ± 13^b

[a] Reference 19.
[b] Without glutaraldehyde.

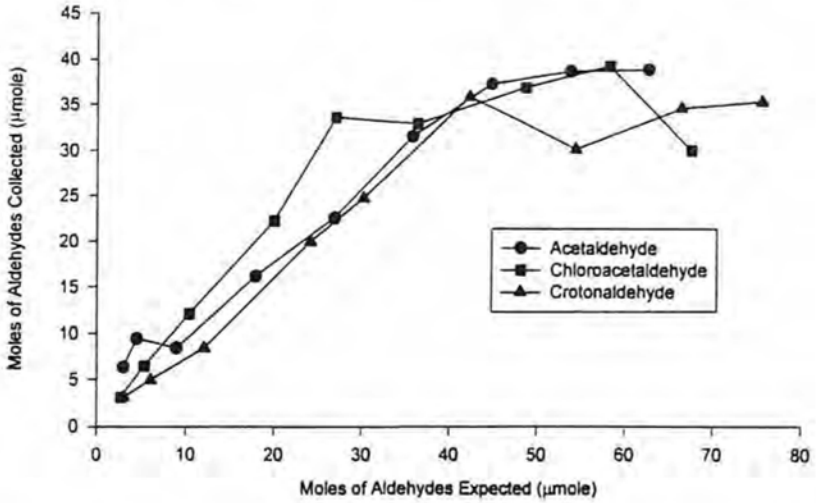

FIGURE 3
Wet spiking capacity tests for acetaldehyde, chloroacetaldehyde, and crotonaldehyde.

Table III shows the results of linear regressions of the aldehyde vapor moles collected versus aldehyde concentration × time curves. The slope is the experimental sampling sampler constant k (equation [4]). Table IV gives the theoretical D and the theoretical k at 25°C and 1 atmosphere from equations (1) and (4). Table IV also shows the experimental k, the experimental D, and sampling efficiencies relative to theoretical.

DISCUSSION

Face velocity, intermittent exposure, temperature, and RH are all important factors which might affect passive sampler performance. These factors were not tested in this research because valeraldehyde and acrolein sampling, which represents the opposite extremes of the PELs, was not affected.[29]

Previous dynamic sampling showed that the absolute recovery for valeraldehyde vapor varied with flow rate, 10 mL/min being better (100 percent) than 50 mL/min (71–85 percent).[19] Flow rate dependence may explain why the sampling efficiency of the passive sampler (79.1 percent relative to equation [4]) was much higher than that for dynamic sampling (23.3 percent at 35 mL/min) for glutaraldehyde. The contact and reaction times were important for the dynamic sampler but not for the passive sampler.

The efficiencies relative to the theoretical sampling constants in Table IV were within ±25% except for chloroacetaldehyde. Why the latter has a higher relative efficiency is unknown. There is no available experimental D for chloroacetaldehyde. Therefore, experimental sampling rates have to be determined for all

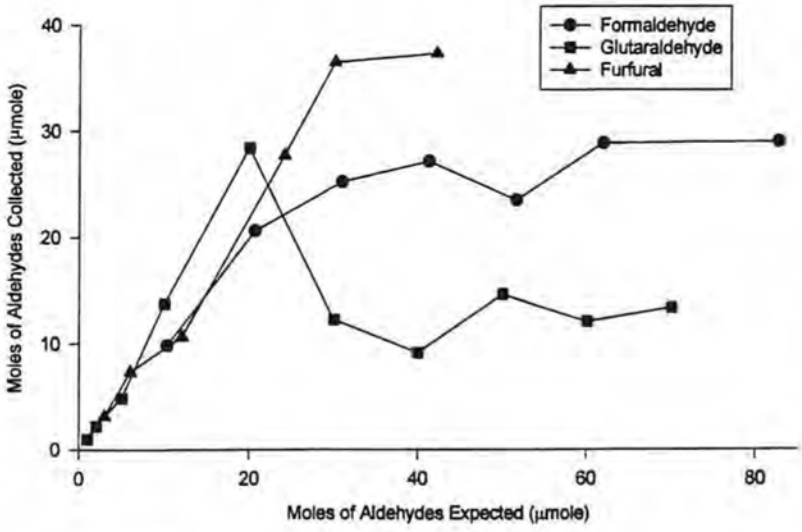

FIGURE 4
Wet spiking capacity tests for formaldehyde, furfural, and glutaraldehyde.

TABLE III
Experimental sampling rates for aldehyde vapor exposures

$$\underset{(\mu\text{mole/mL}) \times \text{min}}{\text{Conc.} \times \text{time}} = k \times \underset{\mu\text{mole}}{\text{moles collected}^{a,b}} + B$$

	k[d] Sampling rate (mL/min)	B (intercept) (μmole/mL) × min	R^2	P
Acetaldhehyde	8.86 ± 0.38	0.02 ± 0.19	0.9963	<0.005
Acrolein[c]	7.73 ± 0.57	0.04 ± 0.12	0.9936	<0.005
Chloroacetaldehyde	11.69 ± 0.32	−0.00036 ± 0.00060	0.9985	<0.001
Crotonaldehyde	7.85 ± 0.19	0.0074 ± 0.0094	0.9988	<0.001
Formaldehyde	9.97 ± 0.055	0.05 ± 0.20	0.9999	<0.001
Furfural	6.47 ± 0.42	0.003 ± 0.011	0.9918	<0.005
Glutaraldehyde	4.46 ± 0.15	0.15 ± 0.16	0.9978	<0.005
Valeraldehyde[c]	4.43 ± 0.19	0.047 ± 0.077	0.9992	<0.001

[a] Moles.
[b] Assumed 100 percent reaction efficiency.
[c] Data from reference 29.
[d] k is defined in equation (4).

vapors of interest because theoretical results are not completely predictive.

The major alternative passive air sampler based on DNPH has been validated for formaldehyde, acetaldehyde, and glutaraldehyde.[25,35,36] The experimental D from the DNPH method and the present PFBHA method for formaldehyde, acetaldehyde, and glutaraldehyde are compared in Table V. There were two different sampler designs for the DNPH method. GMD standard samplers (Hendersonville, Pennsylvania) were used for glutaraldehyde and acetaldehyde sampling, and a filter cassette with a coated 37-mm-diameter filter was used for formaldehyde. The latter accounts for why the theoretical sampling constant for formaldehyde was much higher than the DNPH sampling constants for acetaldehyde and glutaraldehyde. There was no statistical difference at $\alpha = 0.005$ between experimental D values of the same aldehydes for the DNPH and PFBHA methods. This confers credibility to the D values of the other aldehydes.

The DNPH method requires HPLC for analysis which does not distinguish α-hydroxy-carbonyls and the corresponding dicarbonyls like glycolaldehyde/glyoxal, and hydroxyacetone/methylglyoxal, and similar carbonyls such as acrolein, acetone, and propanal. Little attention had been paid to the factors that affect HPLC hydrazone analysis until recently.[37] The absorption spectra vary with pH. Stopped flow HPLC spectra often differ from those of acidified or basic standards, the spectra also varying with the amount of DPNH in standards.[37] There is a pronounced temperature dependence.[37] In addition, the DNPH passive sampler for glutaraldehyde showed a significant temperature effect (lower uptake rate at 12°C and a significantly higher

TABLE IV
Theoretical and experimental sampling constants, diffusion coefficients and relative efficiencies

	Theoretical diffusion coefficient cm^2/sec	Theoretical sampling constant cm^3/min	Experimental diffusion coefficient cm^2/sec	Experimental sampling constant cm^3/min	Relative efficiency[a] (%)
Acetaldehyde	0.128	9.27 ± 0.70	0.1224 ± 0.0052	8.86 ± 0.38	95.6 ± 4.1
Acrolein[b]	0.108	7.84 ± 0.61	0.1067 ± 0.0079	7.73 ± 0.57	98.6 ± 7.3
Chloroacetaldehyde	0.101	7.31 ± 0.55	0.1614 ± 0.0044	11.69 ± 0.32	159.9 ± 4.4
Crotonaldehyde	0.092	6.66 ± 0.50	0.1084 ± 0.0026	7.85 ± 0.19	117.9 ± 2.9
Formaldehyde	0.168	12.16 ± 0.91	0.1377 ± 0.0076	9.97 ± 0.055	81.99 ± 0.45
Furfural	0.083	6.01 ± 0.45	0.0894 ± 0.0058	6.47 ± 0.42	107.7 ± 7.0
Glutaraldehyde	0.078	5.64 ± 0.42	0.0616 ± 0.0079	4.46 ± 0.15	79.1 ± 2.7
Valeraldehyde[b]	0.083	5.95 ± 0.39	0.0612 ± 0.0026	4.43 ± 0.19	74.5 ± 3.2

[a] Relative efficiency (%) $= \frac{\text{experimental sampling constant}}{\text{theoretical sampling constant from equation (4)}} \times 100\%$.
[b] Data from reference 19.

TABLE V
Experimental diffusion coefficients for DNPH and PFBHA methods

	Acetaldehyde		Formaldehyde		Glutaraldehyde	
	DNPH[a]	PFBHA	DNPH[b]	PFBHA	DNPH[c]	PFBHA
Theoretical D[d] (cm^2/sec)	0.123	0.128	0.16	0.168	0.0718	0.078
Theoretical k[e] (cm^3/min)	19.4	9.27 ± 0.70	68	12.16 ± 0.91	13.4	5.64 ± 0.42
Experimental D (cm^2/sec)	0.1084 ± 0.0069	0.1224 ± 0.0052	0.1435 ± 0.0071	0.1377 ± 0.0076	0.0647 ± 0.0082	0.0616 ± 0.0079
Experimental k (cm^3/min)	17.1 ± 1.1	8.86 ± 0.38	61 ± 3	9.970 ± 0.055	11.8 ± 1.5	4.46 ± 0.15

[a]Data from reference 35.
[b]Data from reference 25.
[c]Data from reference 36.
[d]Diffusion coefficient.
[e]Sampling constant.

uptake at 40°C, about 1.5%/°C),[36] even though temperature has no effect on the PFBHA method.[29]

Ozone also interferes with the DNPH-coated silica gel air sampler reaction with air formaldehyde.[38] In contrast, greater sensitivity and resolution are possible for GC/MS and GC/ECD using PFBHA O-oxime derivatives because of the five fluorine atoms in the molecule,[39] thus allowing unknowns to be identified more easily than through the less sensitive liquid chromatography/MS method.[21,40] In addition, reaction efficiencies decrease for dry air relative to humid air for the dynamic DNPH sampling method using coated C_{18} reverse phase cartridges.[26] The latter effect is not shown by Tenax GC- or Tenax TA-coated solid sorbents used in the dynamic[19] or passive sampling modes.[29] Another advantage is the lower detection limit for the PFBHA method (110–200 pg for oxime derivatives)[19] relative to the DNPH method (about 400 ng).[41]

More research should be done on the effects of mixtures for the PFBHA method. In addition, effects of ketones, compounds that also contain a carbonyl group, should be understood because ketones might interfere.

ACKNOWLEDGMENTS

This work was partially presented at the American Industrial Hygiene Conference and Exhibition in Dallas, Texas, May 17–23, 1997, as Abstract 195.

NIOSH/CDCP Grant 5 RO1 OH03120 and the UCLA Center for Occupational and Environmental Health were responsible for financial support of this study.

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APPENDIX 3

OPTIMIZATION OF A SOLID SORBENT DYNAMIC PERSONAL AIR SAMPLING METHOD FOR ALDEHYDES

Yang Shen and Shane S. Que Hee

Department of Environmental Health Sciences and UCLA Center for Occupational and Environmental Health, UCLA School of Public Health, 10833 Le Conte Avenue, Los Angeles CA 90095-1772

Abstract

A solid sorbent personal dynamic air sampling method for aldehydes using chemisorption with 20% (w/w) O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) on Tenax TA has been optimized for several aldehydes. The method for formaldehyde was developed after optimization for valeraldehyde and acrolein. The effects of temperature, intermittent exposure, and flow rate on sampling efficiency were investigated. Vapors of known concentrations were generated in Tedlar gas bags by syringe injection. Aldehyde chemisorption by reaction with 200 mg coated solid sorbent in a Pyrex Tube (7 cm length, 7-mm OD, and 5-mm ID) occurred during dynamic collection with a personal sampling pump operated at 10, 50 or 100 mL/min. The oxime derivatives were 81-87% desorbed by shaking with hexane for 2 minutes or more, resulting in coefficients of variation ranging between 4-7%. Aliquots were then injected for gas chromatographic analysis on a nonpolar capillary column with mass spectrometric or ^{63}Ni electron capture detection. Formaldehyde at 8 ppm-hours equivalent to the permissible exposure limit (PEL) concentration was sampled with a recovery of 94±4% at 50.0 ± 0.5 mL/min. Valeraldehyde and acrolein at their PELs showed no significant differences ($P < 0.05$) in recoveries at different relative humidities (1 and 90 %), temperatures (1, 25, and 40 °C), or intermittent sampling protocol. The sampler capacity at 75% recovery was at a PFBHA:aldehyde molar ratio of 12:1 at 10.0 ± 0.1 mL/min, 17:1 at 50.0 ± 0.5 mL/min, and 26:1 at 100 ± 1 mL/min.

Keywords: air sampling; aldehyde; gas chromatography; PFBHA; solid sorbent; personal sampling; formaldehyde; acrolein; valeraldehyde

Introduction

Aldehydes occur in air as a result of anthropogenic and natural processes,[1,2] incomplete combustion of hydrocarbons, and sunlight-driven photooxidation processes[1-3]. Formaldehyde, acetaldehyde, furfural, and crotonaldehyde are animal carcinogens; formaldehyde is a suspected human carcinogen.[1,4] Aldehydes are mucous membrane irritants and cause pulmonary, skin, eye, and central nervous system effects.[1,3] Growing concern over adverse health effects[1,3,5-7] associated with exposure to formaldehyde and other aldehydes have prompted renewed interest in methods for their measurement. The chromotropic acid method is widely used for formaldehyde[1], and is NIOSH Method 3500[8] and Intersociety Method 116.[9] This method is very sensitive and selective and is capable of measuring ceiling levels of formaldehyde as low as 0.1 ppm.[8] However, the midget impinger technique is not convenient for personal sampling.

Derivatization with 2,4-dinitrophenylhydrazine (DNPH) to form the corresponding hydrazones followed by high performance liquid chromatography (HPLC) and ultraviolet/visible (UV) detection is currently the most popular sampling and analysis technique to determine carbonyl compounds. DNPH air samplers are commercially available.[10,11] EPA protocols T05 and T011 for ambient air carbonyls sampling,[12] Methods IP-6A and IP-6C for indoor air carbonyls sampling,[13] and NIOSH Method 2532 for glutaraldehyde[8] are DNPH-based. The disadvantages of the DNPH method include light sensitivity of the reagent and derivatives, lack of absolute accuracy data,[1] and poor resolution of some DNPH derivatives.[14] Many DNPH hydrazones have low vapor pressures, and decompose partially on high-temperature gas chromatography (GC).

HPLC limits the sensitivity and selectivity of the method, and HPLC/mass spectrometry (MS) is still not developed enough to confirm sensitively unknown aldehyde hydrazones. However, a solid sorbent version does permit personal sampling.

2-Hydroxymethylpiperidine (2-HMP) coated XAD-2 dynamic air sampling is a NIOSH method for workplace personal air sampling and analysis of formaldehyde (Method 2541), acrolein (Method 2501), furfural (Method 2529), valeraldehyde (Method 2536), and aldehyde screening (Method 2539).[8] A nitrogen-specific detector or MS provide sensitive detection after GC. Nonreactive C_3-C_5 aldehydes are not derivatized efficiently, and volatile acids reduce loading capacity.[8]

The O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) derivatization method has been used to detect low molecular weight carbonyl compounds in water using GC/MS.[15-18] A dynamic solid sorbent personal air sampling method for workplace air aldehydes has been developed.[19] Other work with passive sampling for valeraldehyde and acrolein has shown that the coated sorbent has a shelf life of at least 3 months, and a storage stability of at least 6 months after sampling.[20] The GC/electron capture detection (ECD) screening step together with a GC/MS confirmation step make this solid sorbent method more attractive than other available air sampling methods.

The dynamic sampling method developed for several aldehydes did not include formaldehyde.[19] Formaldehyde is the most important of all the aldehydes, since it is ubiquitous, is a animal carcinogen, and is a suspected human carcinogen.[1,4] The

desorption efficiency was optimized as to time and conditions of desorption: the effects of temperature, intermittent exposure, and flow rate were investigated; and the method for formaldehyde assessed using GC/MS and GC-^{63}Ni ECD.

Methods

Reagents

Valeraldehyde (99%), acrolein (97%), and formaldehyde (37% weight/weight (w/w) water solution) were from Aldrich, Milwaukee, Wis. as was the GC internal standard, decafluorobiphenyl. Hexane (Optima), and methanol (Optima) were from Fisher Scientific, Tustin, Los Angeles, CA. PFBHA was from Lancaster Laboratories Inc., Lancaster, PA. Tenax TA (80/100 mesh) was from Alltech Associates, Deerfield, IL. Helium (chromatographic grade), nitrogen (chromatographic grade), and 95% methane/argon (chromatographic grade) were from Alphagaz, Los Angeles, CA.

Equipment

The following were obtained from Fisher Scientific: Pyrex glass tubes 7-mm OD and 5-mm ID, broken into lengths of 7 cm with their ends fire-polished in an air/propane flame; Pyrex glass wool cleaned by hexane Soxhlett extraction; 4-mL Pyrex vials with PTFE-lined caps; Pyrex flasks, volumetric, 10- and 25-mL; pipets, 1-, 2-, 3-, 5-, and 10-mL with pipet bulb; syringes, 10-μL (Hamilton); gas tight syringes, 1-mL (Hamilton); Pyrex beaker, 50-mL; Pyrex flasks, round-bottomed, 250-mL; Pyrex test tube; Soxhlett extraction apparatus; vacuum desiccator; Buchi rotary evaporator; hair dryer (1200 W,

MHT product model 500); and a Mettler AE260 analytical balance. Personal sampling pumps (Pocket Pump) and Tedlar gas bags, 10-L were from SKC Inc., Eighty Four, PA. A Vibro-Graver vibrator was from Burgess Vibrocrafters, North Adams, MA. An M-5 Mini-Buck Calibrator (Buck Scientific, East Norwalk, CT) was used for flow calibrations. A Whatman Zero air generator (Balston Inc.) produced pure air. A dry type bacteriological incubator (Blue M, Blue Island, IL) provided the high temperature conditions (40 °C). An ultrasonic cleaner (Branson Ultrasonics Corporation, Danbury, CT) facilitated ultrasonic agitation. Solid sorbents were Soxhlett-extracted overnight with methanol, and then again with hexane.[19] Glassware was acid-washed.[18]

GC/MS was conducted with a Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, CA). It was equipped with a 30-m x 0.32-mm I.D. DB-1701 chemically bonded fused-silica capillary column (J&W Scientific, Folsom, CA) linked with the 70 eV electron impact ion source of a Hewlett-Packard 5988 quadrupole mass spectrometer equipped with an electron multiplier detector. Samples were injected in the splitless mode with a purge delay of 0.75 minutes, and an injection port temperature of 250 °C. Helium was used as the carrier gas at a flow rate of 3.0 ± 0.3 mL/min. The temperature program was: 105 °C for 0.5 minutes, 105 °C to 220 °C at 10 °C/min, and then holding at 220 °C for 10 minutes. Using the sum of the peak areas of both E- and Z-O-oxime isomers (except for the formaldehyde derivative) to the area of the internal standard (decafluorobiphenyl), linear working ranges were determined as the limit of quantitation of 0.8 ng to 80 ng injected mass of O-oxime for formaldehyde, acrolein, and valeraldehyde O-oxime derivatives, using an injection volume of 2 μL. The working

ranges were equivalent to 0.16 μg to 16 μg formaldehyde per sample, 0.27 μg to 27 μg acrolein per sample, and 0.37 μg to 3.7 μg valeraldehyde per sample. With a sampling volume of 3 L, the working range corresponded to 0.04 ppm to 4 ppm of these aldehydes.

The same column and temperature conditions were used for analysis by Hewlett-Packard 5890 GC/^{63}Ni-ECD at 5 % methane/argon flow of 3.0±0.4 mL/min, and detector temperature of 250 °C. The signal was displayed by a Hewlett-Packard 3396 integrator.

Sampler Preparation

Before coating, solid sorbents were cleaned by Soxhlett extraction with methanol overnight, and then with hexane overnight. The solid sorbent was dried in an oven at 80 °C for 2 hours, and then placed in a vacuum desiccator until constant weight (usually 2 days) was obtained. The PFBHA coating of 20% weight/weight (w/w) was chosen to maximize the mole ratio of PFBHA to aldehyde, the capacity of the sampling tube, and to follow the lead of other investigators.[19] Thus 0.75 g (3 mmoles) PFBHA was dissolved in 25-mL methanol solution, and shaken. The solution was poured into a 250-mL round bottom flask that contained 3 g of Tenax TA. The solvent was removed by rotary evaporation at 50 °C for 1 hour, and placed in a vacuum desiccator at room temperature until constant weight was attained. A 200-mg weight of the coated solid sorbent was packed into a Pyrex tube (7-cm long, 7-mm OD, 5-mm ID) by a vibrator, and contained between two 5-mm layers of Pyrex wool cleaned by the same method as the sorbent.

Test Atmosphere Preparation

Syringe injection of a known volume of aldehyde of known purity into a Tedlar gas bag generated the test atmosphere.[19, 21] A hair dryer ensured volatilization and mixing. After emptying (vacuum) and filling three times, the third gas bag was retained for testing. Different relative humidities (RH) were generated by syringe injection of calculated volumes of water.[19] The RH was either 1% or 90%.[19] For acrolein and formaldehyde experiments, a concentrated atmosphere was generated first. A gas-tight syringe allowed transfer of a known volume of concentrated vapor into a known volume of air in another Tedlar gas bag to obtain the desired concentration. The concentrations for formaldehyde for one hour sampling were 4, 8, or 32 times its OSHA PEL, equivalent respectively to 0.5, 1, or 4 times PEL ppm-hour exposure conditions over an exposure period of 8-hours. All sampling experiments were done in triplicate.

Synthesis of PFBHA O-Oximes

Pure O-oximes had to be synthesized to obtain absolute sampling efficiency since they are not commercially available. The O-oximes of valeraldehyde and acrolein had been made previously.[18, 19] Standardization of commercial formalin[8] resulted in a concentration of formaldehyde of 0.31 ± 0.01 g/mL. Formaldehyde water solution (97 μL; 1.00 mmole) was added in a test tube containing 5 mL distilled water. A mass of 249.5 mg (1.00 mmole) of PFBHA was dissolved in 10 mL distilled water, and the solution then poured into the diluted formalin. The mixture was shaken 15 minutes, heated in a microwave oven until the first bubbles appeared, and cooled in an ice bath for 30 minutes. After centrifugation for 5 minutes, the tube was placed in the ice bath again.

As much of the upper layer of water as possible was removed by Pasteur pipet. A volume of 1 mL hexane was added to extract the derivative, and centrifuged to separate the two layers. The hexane layer was retained, and the extraction procedure repeated three more times. The solvent was evaporated under nitrogen gas at room temperature. The vial containing the liquid residual was placed in the vacuum desiccator until constant weight was attained. The purity of synthesized formaldehyde PFBHA O-oxime by GC/MS was 99±1%. The yield was 87±7%.

Optimization of Aldehyde O-Oxime Desorption

The previous desorption method[19] recommended standing for 2 hours with occasional shaking after liquid or vapor spiking. Both liquid spiking and vapor spiking were again evaluated. Valeraldehyde, a relatively nontoxic aldehyde with the highest PEL (50 ppm), and acrolein with the lowest PEL (0.1 ppm) were selected as the screening test chemicals. For liquid spiking, 1 μL of valeraldehyde was spiked onto 200 mg 20% (w/w) PFBHA-coated Tenax TA in a sampling tube. Triplicate samples were spiked, and allowed to stand overnight at room temperature. In addition, 1 μL of valeradehyde or 2 μL of 1/1000 volume/volume (v/v) acrolein/methanol solution was spiked onto a sampling tube containing 200 mg 20% (w/w) PFBHA-coated Tenax TA. Clean air was drawn through the tube at 50 mL/min for 2 hours. All the samples were desorbed with 3 mL hexane in a 4-mL glass vial with PTFE-lined cap. The different desorption procedures were: 1 minute and 2 minutes ultrasonic agitation; 2 minutes manual agitation; 1 hour desorption with manual agitation (every 15 minutes); 2 hours desorption without agitation; and 2 hours desorption with manual agitation every 30 minutes.

Sampling Procedure

The flow rates utilized were from 9.5 to 10.5 mL/min with an average of 10.00 ± 0.08 mL/min; from 45 to 55 mL/min with an average of 50 ± 1 mL/min; and from 95 to 105 mL/min with an average of 100 ± 1 mL/min. All air volumes were referenced to 25 °C and 760 mm Hg. PTFE Teflon tubing (5-mm I.D.) was used to connect the Tedlar air bag and sampling tube via a small piece of Tygon tubing as a butt-to-butt connector. The personal sampling pump was attached to the sampling tube with Tygon tubing.

Temperature

Temperature influences the equilibrium and rates of reactions.[22] Both high and low temperature may influence the equilibrium of the chemisorption reaction, and sampling efficiency. Three temperatures were tested. The low temperature of 7±1 °C was achieved in a walk-in refrigerator. The temperature of 25 ± 1 °C was room temperature. The high temperature of 40 ± 2 °C was obtained inside a calibrated incubator.

Intermittent Exposures

In the workplace, vapor exposure concentrations always change. Intermittent exposure often limits the use of physical adsorption for the air sampling of volatile compounds due to back-adsorption in passive samplers, and migration during dynamic sampling and during storage. Chemisorption with PFBHA produces a less volatile derivative that has high affinity for the solid sorbent, thus concentrating it, and preventing volatilization and minimizing migration. Different intermittent exposure situations were tested using valeraldehyde as a test compound. Those situations included a 1-hour peak exposure of 8

times PEL at the beginning of a 8-hour exposure or 1-hour peak exposure of 8 times the PEL at the end of a 8-hour exposure.

Capacity

Using valeraldehyde as reference compound, the capacities of the sampler at different flow rates of 10, 50, and 100 mL/min were determined in terms of the PFBHA:aldehyde molar ratio. Air sampling was conducted using valeraldehyde vapor of different ppm-hour exposure levels.

Results and Discussion

Table 1 shows the results of the recovery optimization for the valeraldehyde liquid spiking experiment. Except for the desorption conditions of 1 minute ultrasonic agitation, and 2 hours desorption without agitation, there are no significant differences at $p \leq 0.05$ between the recoveries of all the other desorption conditions. The latter recoveries range from 81 to 87%, with coefficients of variation (CV) of 4 to 7%. Agitation affects desorption efficiency, and a critical desorption duration of at least 2 minutes with sample agitation is necessary.

Table 2 gives the results of the valeraldehyde vapor spiking experiment. The recoveries range from 84 to 93%, with CVs of 2 to 10%. There are no significant differences ($p \leq 0.05$) among recoveries. Comparing the data of Table 1 and 2, liquid spiking and vapor spiking are equivalent and exceed 75% recovery[8]. Thus liquid spiking suffices for derivative formation, an important attribute since there are no commercial PFBHA O-oxime standards. Table 3 depicts the results of acrolein vapor spiking. The

recoveries range from 101 to 104%, with CVs of 3 to 10%. There were no significant differences ($p \leq 0.05$) among recoveries.

The data of Table 1 through 3 also show that the desorption duration can be shortened to 2 minutes with manual agitation or ultrasonic agitation. This will speed analyses compared with using the 2-hour desorption duration used previously.[19]

The influence of flow rate on capacity for valeraldehyde is given in Figure 1. In general, for the same flow rate, as the PFBHA:aldehyde molar ratio decreased, the sampling efficiencies (mass of O-oxime desorbed relative to that at saturation) decreased due to breakthrough below a critical ratio. The shape of the sampling efficiency versus PFBHA:aldehyde molar ratio curves has a broad plateau near 75% saturation except at the lowest flow rate. The critical PFBHA:aldehyde ratio at 75% capacity was 12:1 at 10 mL/min, 17:1 at 50 mL/min, and 28:1 at 100 mL/min. Breakthrough will occur for sampling below these molar ratios at these flow rates. As the flow rate increased, sampler capacity decreased. Sufficient contact time at these flow rates is necessary for the reaction to be complete. While the theoretical capacity was 805 μmole, not all of this was actually available.

After logarithmic transformation, the relationship between the sampler capacity in terms of PFBHA:aldehyde molar ratio and flow rate was linear with $P=0.0243$ as shown in Figure 2. The latter allows prediction of the sampler capacity in terms of PFBHA: aldehyde molar ratio between 10 to 100 mL/min, the practical flow rate range for

occupational sampling. Thus, whereas low concentrations can be sampled at the higher flow rates, high concentrations must be sampled at lower flow rates to prevent breakthrough. Probably, the high flow should be used first in the field, and depending on the results, the low flow then used if indicated.

The relationship between the logarithm of the critical PFBHA:aldehyde molar ratio at 75% bed capacity and flow rate was:

$$\log \text{ratio} = (0.00410 \pm 0.00016)\ \text{flow rate} + (1.034 \pm 0.010) \qquad (1)$$

The intercept of 1.034 for equation (1) is significantly different from 0 and from 1 at $P \leq 0.05$. The capacity ratio at flow rate of zero is significantly different from 1. For flow rate $\rightarrow$ 0, log capacity ratio $\rightarrow$ 1.034. Assuming the same relative standard deviation as the intercept, the log ratio of 1.0 has a standard deviation of 0.010. After back transformation, the ratio is 10.8 with a range of 10.7 to 11.2. This implies that the coated sorbent might be suitable for passive sampling. For example, if 200 mg 20% coated sorbent were available for passive sampling, the theoretical aldehyde capacity at 75% that could be sampled is: moles of PFBHA/limiting ratio = 160 μmoles / 10.8 = 14.8 μmoles with a range of 14.3 to 15.0 μmoles. Other work has shown that passive sampling is indeed possible.[20]

Table 4 gives the results of valeraldehyde sampling at different RHs, temperatures, and flow rates. All the sampling efficiencies are higher than 75% with CVs of 3 to 8%. The

recoveries exceeded 90% when breakthrough did not occur. The lower recoveries at 50 mL/min were because of breakthrough. Temperature, RH, and the combination of temperature and RH did not influence the chemisorption and desorption efficiencies.

The results of the acrolein sampling temperature test at a concentration of 0.25 mg/m^3, RH of 1%, and at flow rate of 50 mL/min for a sample volume of 5 L are shown in Table 5. The recoveries all exceed 96 %. Temperature had no influence on the recovery. This agrees with the results for valeraldehyde in Table 4.

The results of the intermittent exposure test for valeraldehyde at 10 mL/min are provided in Table 6. The data show that intermittent exposure is not a factor that influences sampling efficiency, the recoveries at this high air concentration being between 76-79 %. After chemisorption, the derivative remained on the solid sorbent, and did not break through. This may not be the case for methods based on physical adsorption, where analyte migration and breakthrough occur.[8]

Table 7 shows the results of formaldehyde experiments at different temperatures at 50 mL/min for different concentrations ranging from 4.8-38.4 mg/m^3. The sampling efficiencies are from 90 to 106%, with CVs of 4 to 10%. There are no significant differences ($p \leq 0.05$) among the O-oxime recoveries. Temperature has no influence on formaldehyde sampling efficiency. Thus, formaldehyde sampling has the same characteristics as those for valeraldehyde and acrolein.[19]

The above results were not intended to fulfill a NIOSH validation protocol where a minimum of six replicates have to be evaluated at each of at least three different concentrations for vapor spikings, and at least four different loadings (0.1, 0.5, 1.0 and 2.0 the target ppm-hours) for liquid spikings. The NIOSH recovery criteria in the fourth edition of the NIOSH Manual of Analytical Methods of 1994[8] are explained thus: "For the 0.5, 1, and 2 times the target concentration, recoveries of greater than or equal to 90% are preferred although recoveries greater than or equal to 75% may be acceptable." Using the 90% recovery criterion, the optimized method recovery for 4.8-38.4 mg/m^3 formaldehyde (Table 7) and 0.25 mg/m^3 acrolein (Table 5) sampled at 50 mL/min, and for 162 mg/m^3 valeraldehyde sampled at 10 mL/min are acceptable for total volumes of 1-5 L between 7-40 °C and 1-90% RH. NIOSH mentions no temperature test for dynamic samplers though it recommends such testing for passive samplers at about 10, 25, and 40 °C. Valeraldehyde of 162 mg/m^3 concentration sampled at 50 mL/min (Table 4), and of 1408 mg/m^3 sampled at 10 mL/min (Table 6) do not meet the 90% recovery criterion but still exceed 75% recovery. The NIOSH collected sample stability criterion of 30 days is met, but there is no shelf life minimum criterion. Investigation of the dependence of dynamic sampler capacity on flow rate is not mentioned either.

The PFBHA method can utilize GC/ECD as a screening method or GC/MS as a confirmatory method. The latter using m/z 181 selective ion monitoring is a major advantage in identifying and quantifying unknown aldehydes. Both ECD and MS detectors can detect the PFBHA O-oxime at the pg level which is much more sensitive than HPLC/UV used in the DNPH method. The working air concentration range of 40

ppb to 4,000 ppb for a 3-L sample is sufficient for workplace purposes, higher concentrations being accessible through dilution, and also lower ones to ultimately about 10 ppt. The ECD is also less expensive and more commonly found in the laboratory than the nitrogen detector used for screening purposes in the NIOSH method.

Conclusions

The PFBHA coated Tenax-TA dynamic air sampling method has been optimized for the air sampling of valeraldehyde, acrolein, and formaldehyde. Formaldehyde at 8 ppm-hours relative to the PEL, 0.5 PEL and 4 PEL concentration can be sampled with a recovery higher than 90%. Temperature, relative humidity, and intermittent exposure do not affect the sampling efficiency. The recommended desorption procedure is 2 min ultrasonic agitation, or 2 min manual agitation. The capacities for this sampler are PFBHA:aldehyde molar ratio of 12:1 at a sampling rate of 10 mL/min, 17:1 at 50 mL/min, and 28:1 at 100 mL/min. The lower the concentration, the higher the sampling flow rate that can be used.

Recommendations

The optimized PFBHA coated Tenax-TA dynamic air sampling method can be used in the sampling of workplace air formaldehyde, acrolein, and valeraldehyde using an analytical technique (GC/ECD or GC/MS) that is already widely used, compared to a nitrogen-specific GC detector for the NIOSH method for aldehydes, or that is more selective and accurate than the HPLC/UV method for aldehydes of EPA. The flow rate can be set at 10 mL/min or 50 mL/min depending on the aldehyde concentration.

Acknowledgments

This project was supported by Grant 1 R01 OH03120 from the National Institute for Occupational Safety and Health of the Centers for Disease Control and Prevention. Partial support also came from the UCLA Center for Occupational and Environmental Health and the UCLA Center for Environmental Risk Reduction. Dr. Yu-Wen Lin and Dr. Shih-Wei Tsai are thanked for their assistance. This research was presented partially at the American Industrial Hygiene Association Conference and Exhibition, Atlanta, Georgia, May 11-15, 1998, as Abstract 211.

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Captions for Figures

Figure 1. Sampler Capacity Efficiency Test Relative to the PFBHA/Aldehyde Molar Ratio at Different Flow Rates at 25 °C.

Figure 2. Dependence of Log (PFBHA/Aldehyde Molar Ratio at 75% Capacity) on Flow Rate at 25 °C.

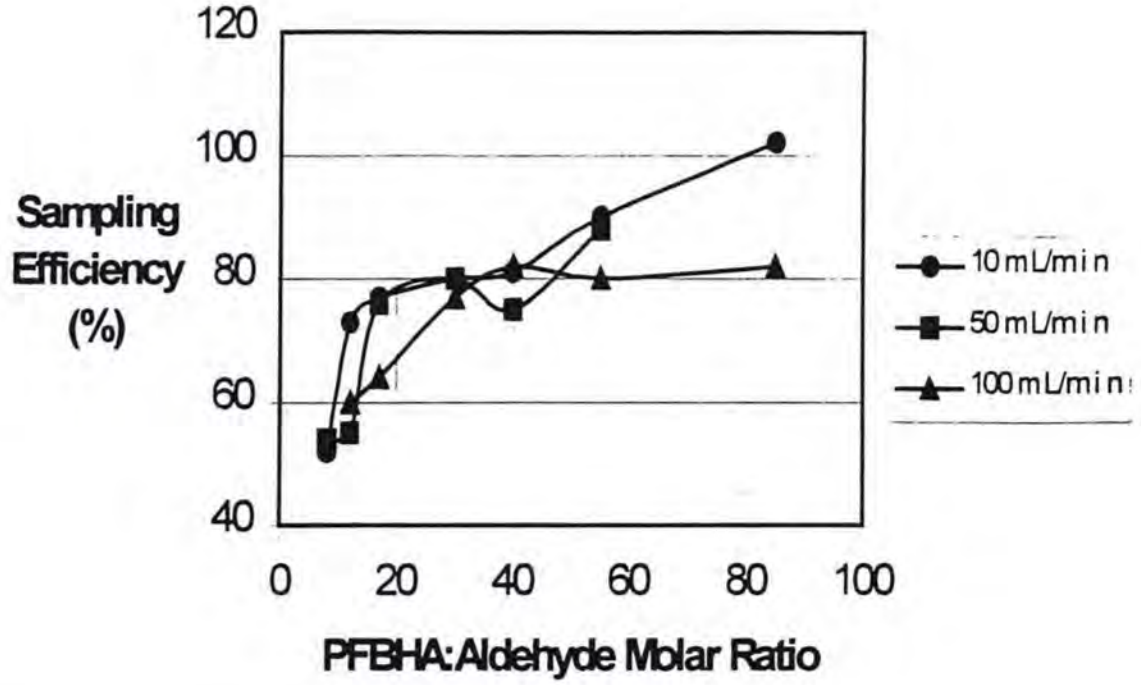
120
100
80
60
40
Sampling Efficiency (%)
0
20
40
60
80
100
PFBHA:Aldehyde Molar Ratio
10 mL/min
50 mL/min
100 mL/min

Figure 1.

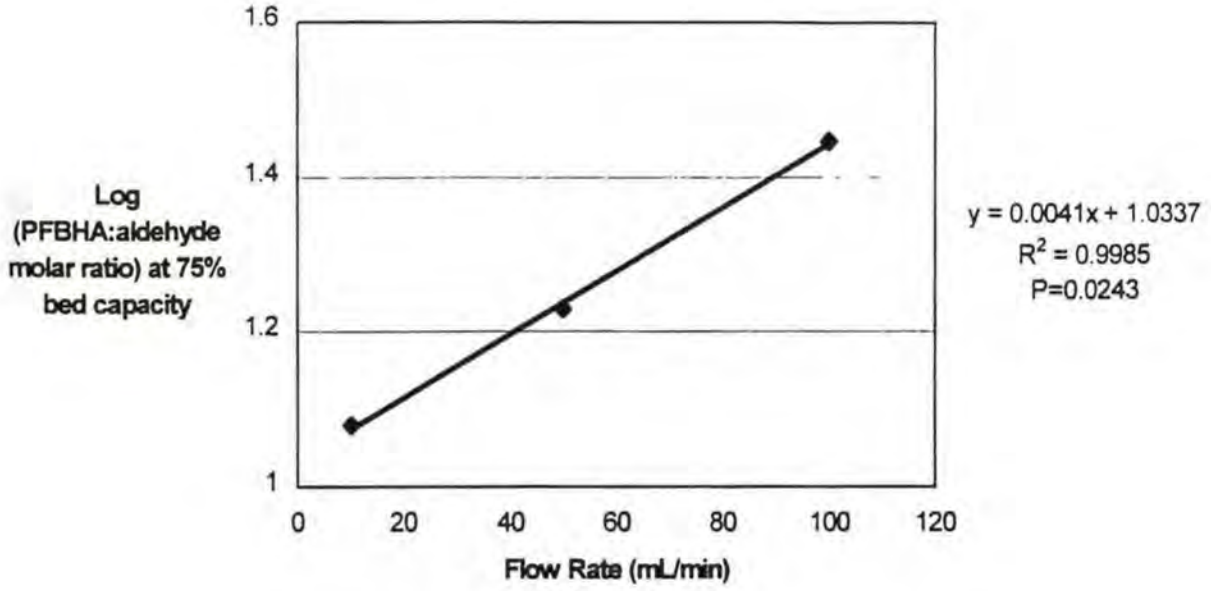
Log (PFBHA:aldehyde molar ratio) at 75% bed capacity
1.6
1.4
1.2
1
0
20
40
60
80
100
120
Flow Rate (mL/min)
y = 0.0041x + 1.0337
R2 = 0.9985
P=0.0243

Figure 2.

Table 1. Optimization of Valeraldehyde Liquid Spiking Recovery as the O-Oxime at 25 °C.

Desorption Conditions		Recovery	CV[b]
Method	Duration (min)	(%)	(%)
Ultrasonic	1	67[a]	1
Ultrasonic	2	81	5
Manual	2	83	4
Manual	60 (15-min intervals)	86	7
Manual	120	87	6
Static	120 (30-min intervals)	55[a]	14

a. Significantly different at $P \leq 0.05$.

b. Coefficient of variation.

Table 2. Optimization of Valeraldehyde Vapor Spiking Recovery as the O-Oxime at 25 °C.

	Desorption Condition	Recovery	CV
Method	Duration (min)	(%)*	(%)
Ultrasonic	2	85	7
Manual	2	84	10
Manual	60 (15-min intervals)	90	10
Manual	120 (30-min intervals)	93	2
	Range	84-93	2-10

* No significant difference at $P \leq 0.05$.

Table 3. Optimization of Acrolein Vapor Spiking Recovery as the O-Oxime at 25 °C.

Desorption Condition		Recovery	CV
Method	Duration (min)	(%)*	(%)*
Ultrasonic	2	101	10
Manual	2	100	3
Manual	60 (15-min intervals)	104	5
Manual	120 (30-min intervals)	101	8
	Range	100-104	3-10

* No significant difference at $P \leq 0.05$.

Table 4. Sampling Efficiencies for Valeraldehyde at 162 mg/m^3 at Different Conditions of Relative Humidity (RH), Temperature (Temp.), Flow Rate, and Sample Volume.

Temp. (°C)	RH (%)	Flow Rate (mL/min)	Sample Volume (L)	Recovery (%)	CV (%)
25	1	50	5	76*	3
7	1	50	5	82*	7
40	1	50	5	76*	3
25	1	10	1	102	4
7	1	10	1	97	5
40	1	10	1	99	6
25	90	10	1	97	7
7	90	10	1	93	8
40	90	10	1	101	5
Range: 7-40	1-90	10-50	1-5	76-102	3-8

* Significantly different at $P \leq 0.05$ from the 10 mL/min data due to breakthrough.

Table 5. Sampling Efficiencies for Acrolein at 0.25 mg/m^3, 50 mL/min, 5 L Sampling Volume, and RH of 1% at Different Temperatures.

Temperature (°C)	Recovery (%)*	CV (%)
25	101	2
7	96	4
40	97	6
Range: 7-40	96-101	2-6

* No significant difference at P≤ 0.05.

Table 6. Intermittent Exposure Results for Valeraldehyde at 1408 mg/m^3 at 10 mL/min at 25 °C.

Exposure Condition	Recovery (%)*	CV (%)
1. Continuous for 1 hour	77	3
2. 1 Hour exposure / 7 hours zero air	76	3
3. 7 Hours zero air / 1 hour exposure	79	5
Range	76-79	3-5

* No significant difference at P≤0.05.

Table 7. Sampling Efficiencies for Formaldehyde with the Optimized Method at 50 mL/min, Different Air Concentrations, Temperatures, and Sample Volume.

Vapor Conc. (mg/m^3)	Temperature (^oC)	Sample Volume (L)	Recovery (%)	CV (%)
9.6	25	3	94	4
9.6	40	3	90	6
9.6	7	3	102	8
4.8	25	3	106	10
38.4	25	3	97	9
Range: 4.8-38.4	7-40	3	90-106	4-10

* No significant differences at $P \leq 0.05$.

APPENDIX 4

REGULATED WORKPLACE KETONES

AND THEIR INTERFERENCE

IN THE PFBHA METHOD FOR ALDEHYDES

YU-WEN LIN AND SHANE S. QUE HEE*

**Department of Environmental Health Sciences and
UCLA Center for Occupational and Environmental
Health
UCLA School of Public Health
10833 Le Conte Avenue
Los Angeles CA 90095-1772**

NIOSH/CDCP Grant 5 RO1 OH03120 and the UCLA Center for Occupational and Environmental Health were responsible for financial support

* To whom correspondence should be sent

REGULATED WORKPLACE KETONES AND THEIR INTERFERENCE IN THE PFBHA METHOD FOR ALDEHYDES

ABSTRACT

Ketones are the major positive interferences for an aldehyde dynamic air sampler that consists of 200-mg 20 % (w/w) O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) on Tenax TA contained in a Pyrex tube 7-mm OD, 5-mm ID and 70-mm in length, that utilizes a personal battery-powered pump at 10-50 mL/min. The ketone O-oxime derivatives were synthesized to allow absolute quantitation of O-oximes formed during sampling. Wet spiking allowed ketone recoveries to be found. Ketone vapors of known concentrations were generated statically in Tedlar gas bags. The O-oximes were desorbed with hexane, and an aliquot injected for gas chromatographic analysis on a nonpolar capillary column with mass spectrometric or electron capture detection. Gas phase recoveries up to 200 ppm-hour loadings exceeded 75% at 25 °C for chloroacetone, cyclohexanone, diacetone alcohol, diethyl ketone, dipropyl ketone, ethyl butyl ketone, methyl amyl ketone, methyl butyl ketone, 2-methylcyclohexanone, methyl ethyl ketone, methyl *iso*butyl ketone, methyl *iso*propyl ketone, and methyl propyl ketone. The recoveries for acetophenone, 2-chloroacetophenone, and ethyl amyl ketone were lower than 75%, and were caused by steric hindrance. Sampling for both aldehydes and ketones is recommended at 10 mL/min for TLV concentrations.

Keywords: dynamic sampling; ketone; solid sorbent sampling; adsorption; oxime; gas chromatography

Ketones (R_1-(C=O)-R_2 where R_1 and R_2 are alkyl, aromatic, or alicyclic functional groups) are widely used industrial chemicals. They are used as solvents, chemical intermediates, cleaning fluids, dewaxers, and reaction enhancers, as well as in paints, hydraulic fluids, cleaning fluids, inks, pharmaceuticals, cosmetics, and dopes[1,2]. Probably the most prevalent exposures in workplaces are during painting and lacquering operations, in paint factories, and in chemical laboratories during solvent dispensing. Commercially important ketones include acetone, diacetone, methyl ethyl ketone, methyl propyl ketone, and methyl isobutyl ketone[2]. Ketones are also environmental products of photooxidation. For example, methyl ethyl ketone can be produced in outdoor air by the photooxidation of such air pollutants as butane and other hydrocarbons[3]. Methyl ethyl ketone has also been found in drinking water and surface waters[4], and is also produced as a product of metabolism[5]. Ketones are emitted as products of bacterial spoilage[6], oxidative combustion[7], and are important markers of lipid peroxidation, metabolic status, and diabetic status[8], as measured through breath sampling[5]. Water-soluble ketones dehydrate and then abrade the skin after contact, allowing enhanced skin permeation of other exposing chemicals.

Ketones are mucous membrane irritants and activate the trigeminal nerve endings in the eyes and nose ("sensory irritation"), but are not as potent as their closely related aldehyde analogs of the same number of carbon atoms. Overexposure can cause narcosis, headache, nausea, light-headedness, dizziness, and incoordination. Methyl

butyl ketone is oxidized to the same neurotoxic metabolite (2,4-hexanedione) as is *n*-hexane, and peripheral and central neuropathy are caused in rats after TWA exposure to 1,300 ppm[2].

Methods for personal sampling of ketone vapors usually involve dynamic air sampling with solid sorbents[9]. NIOSH recommends several methods like charcoal tube sampling for acetone, cyclohexanone, diisobutyl ketone, 2-hexanone, methyl isobutyl ketone, and 2-pentanone[10]. However, CS_2 desorption of the more nonpolar ketones on charcoal tubes is inefficient. A desorbing mixture of 1% methanol in CS_2 improves desorption of camphor, mesityl oxide, 5-methyl-3-heptanone, methyl-(*n*-amyl) ketone, and ethyl butyl ketone[11]. Methyl ethyl ketone is sampled on beaded carbon before desorption by CS_2 [12]. 2-, 3- ,and 4- methyl cyclohexanone are sampled on Porapak Q, desorbed with acetone, and analyzed by GC[13]. Thermal desorption from graphitized carbon and carbon molecular sieves is used for ppb concentrations of ketone vapors [14].

The 2,4-dinitrophenylhydrazine (2,4-DNPH) solid sorbent method is recommended by the United States Environmental Protection Agency[15] to determine aldehydes and ketones in ambient air. The 2,4-DNPH method potentially allows relatively selective quantitation of different aldehydes and ketones through high performance liquid chromatography (HPLC)/ultraviolet detection (UVD) of their hydrazones but not by

GC since many hydrazones decompose at high temperatures[9]. 2,4-DNPH does not react quantitatively with conjugated aliphatic aldehydes, can be light sensitive, is prone to ozone interference, and variable recoveries occur on liquid spiking[9]. Some passive samplers have been developed for the lower molecular weight aldehydes and ketones based on liquid systems[16,17]. Solid sorbent DNPH passive samplers are available[18-21].

O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) has been used to analyze aldehydes in ozonated water[22] because of its fast quantitative reaction to form O-oximes that can be detected at the picogram (pg) level by gas chromatography/mass spectrometry (GC/MS) and gas chromatography/electron capture detection (GC/ECD)[23]. The PFBHA method has been used to chemisorb aldehyde vapors by dynamic sampling[24,25], and by passive sampling[26,27]. In those studies, relative humidity RH (3 to 79 %), temperature (4 to 48 °C), intermittent exposures, shelf life (at least 3 months), and storage stability (at least 6 months) were shown to have no effects on aldehyde O-oxime recoveries, and thus the method shows promise. The present study extends the PFBHA dynamic sampling method for aldehydes to selected regulated ketones, the major positive interferences of the aldehydes.

EXPERIMENTAL METHODS

Materials

The ketones from Aldrich, Milwaukee, WI, were: acetophenone (99%), butyl ethyl ketone or 3-heptanone (98%), chloroacetone or chloro-2-propanone (95%), 2-chloroacetophenone or phenacyl chloride (99%), diacetone alcohol or 4-hydroxy-4-methyl-2-pentanone (99%), di propyl ketone or 4-heptanone (98%), cyclohexanone (99.8%), diethyl ketone or 3-pentanone (99+%), ethyl amyl ketone or 3-octanone (98+%), methyl amyl ketone or 2-heptanone (98%), methyl butyl ketone or 2-hexanone (98%), 2-methylcyclohexanone (99%), methyl ethyl ketone or 2-butanone (99+%), methyl isobutyl ketone or 4-methyl-2-pentanone (99.5+%), methyl isopropyl ketone or 3-methyl 2-butanone (99%), and methyl propyl ketone or 2-pentanone (99.5%). Internal standard decafluorobiphenyl (99%) was also from Aldrich. Hexane (Optima), methanol (Optima), nitric acid, activated charcoal, molecular sieves, and indicating Drierite were from Fisher Scientific, Tustin, CA. O-(2,3,4,5,6-Pentafluorobenzyl)hydroxylamine hydrochloride(PFBHA) was from Lancaster Laboratories, Lancaster, PA. Tenax TA (80/100 mesh) was from Alltech Associates, Deerfield, IL. Chromatographic grade helium, nitrogen, and 5% methane/argon were from Alphagaz, Los Angeles, CA.

Equipment

Pyrex tubing (7-mm OD, 5-mm ID), Pyrex glass wool, 4-mL Kimble vials with

PTFE-lined screw caps, 10-μL Hamilton syringes for chromatographic injections, gas-tight Hamilton syringes, Soxhlett-extraction apparatus, calibrated temperature/ relative humidity (RH) meter/recorder, and a hair dryer to vaporize liquids and homogenize atmospheres in gas bags were from Fisher Scientific. Pocketpumps (Model No. 210-1002), rotameters, and Tedlar gas bags were from SKC West, Fullerton CA. A Whatman Zero Air generator was from Balston, Haverhill, MA. A M-5 Mini-Buck calibrator for flow rate measurement was from Buck Scientific, East Norwalk, CT. A Goldstar Multiwave microwave oven facilitated O-oxime syntheses.

GC/MS was done with a Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, Calif.) equipped with a 30-m × 0.32-mm ID DB-1701 chemically bonded (1-μm thick film) fused-silica capillary column. The temperature for the injector, and link was 250 °C. The column temperature program was: solvent delay 5 min at 105 °C, 105 °C for 0.5 minutes, 105 °C to 200 °C at 10 °C/minute, and holding then until all peaks eluted. The Hewlett-Packard 5988A quadrupole positive ion electron impact mass spectrometer had an electron multiplier detector, and the 70-_eV ion source temperature was 250 °C. Selective ion monitoring (SIM) used m/z 181 and total ion monitoring (TIC) utilized m/z 50-500. The areas of both *E*- and *Z*- isomers were utilized for quantitations.

The same column, temperature, and peak quantitation conditions were used for

Hewlett-Packard 5890 capillary GC/^{63}Ni-electron capture detection (ECD) with 5% methane/argon carrier flow of 3.0±0.4 mL/min. The detector temperature was 250 °C. The flows for the septum purge, anode, and make-up carrier gas were 3.0±0.2, 4.0±0.3, and 40±3 mL/min, respectively. The signal was visualized with a Hewlett-Packard 3396 integrator. As for GC/MS, the injection volume was 2 μL.

Methods

After the synthesis of the new pure ketone O-oximes of PFBHA to provide absolute quantitations, the sampling tubes were prepared. Spiked ketones (neat or in methanol) at guideline equivalent mass levels allowed calculation of wet spiking recoveries. Known ketone vapor concentrations generated in Tedlar gas bags allowed determination of vapor recoveries when a known volume was sampled.

Synthesis of PFBHA O-Oximes

The PFBHA O-oximes are not commercially available. They were synthesized in triplicate by methods detailed elsewhere[22,24,28]. All the PFBHA O-oximes synthesized for this work in Table 1 are new, except those for methyl amyl ketone, acetophenone, 2-methylcyclohexanone, and acetone, these being synthesized before[28]. Each derivative in hexane at a total injection mass of about 1 μg in 2 μL was subjected to GC/MS investigation in the total ion current (TIC) mode for purity. The areas of both *E*- and *Z*-isomers of the PFBHA O-oximes were utilized for

quantitation purposes. This TIC-GC/MS corrected for the presence of pentafluorobenzaldehyde, pentafluorobenzyl alcohol, excess PFBHA in the O-oximes, any other aldehydes, and other peaks not attributable to the reagents and solvents[22]. Yields were corrected for GC/MS purity. Other spectroscopic criteria of purity (ultraviolet, infrared, mass spectra, and ^{1}H- and ^{13}C- nuclear magnetic resonance) for the 4 ketones already synthesized before this study are also available[28]. Quantitations utilized the selected ion monitoring (SIM) mode at m/z 181. GC/^{63}Ni-ECD was also used with the same capillary column, temperature program, and column flow rates.

Sampler Preparation. The Tenax TA and glass wool were separately Soxhlett-extracted overnight with methanol, and then hexane. Both were dried to constant weight in a vacuum desiccator containing indicating Drierite. PFBHA (0.7500 g) was dissolved in 25 mL methanol and added to 3.0 g Tenax TA in a preweighed 250-ml 24/40 pyrex ground glass round bottom flask to produce a 20% w/w PFBHA coating. The methanol was removed by rotary/vacuum evaporation at 55 °C until the solid flowed. The container was then left in a vacuum desiccator until constant weight of the solid product.

Pyrex tubing was cut into 70-mm lengths, the ends fire-polished, acid-cleaned, and dried. After inserting a 5.0-mm glass wool plug at one end, 200 mg of the coated

solid sorbent was packed uniformly into the tube using the vibrator. Once the upper distance of the sorbent top to the end of the tube was constant on continued vibration, another 5.0-mm glass wool plug was inserted at the upper surface. The ends were capped for storage in a vacuum desiccator at room temperature.

Desorption Efficiency. Specific volumes of ketone or ketone/methanol solution containing known weights of ketone equivalent to the target TLV-TWA ppm-hour (except for chloroacetone which was the only ketone examined that had a Ceiling Limit where 0.25 x Ceiling Limit ppm was spiked) were spiked onto the tubes in triplicate as shown in Table 2. After standing overnight, desorption was done with 3-mL hexane in a capped scintillation vial by 2-min manual agitation at 25 °C[25].

Sampling Efficiency. Known concentrations of specific ketones were prepared in triplicate in 10-L Tedlar gas bags statically by injecting known volumes[24] of ketones into purified compressed air of 1% RH[25-27], and heating the mixture with a hair dryer to facilitate complete ketone evaporation and mixing. The low RH was chosen as the most adverse RH condition since the aqueous solution reaction was known to be quantitative from the synthesis of the PFBHA O-oximes of the ketones at the beginning of the present study. Previous work gave similar results for the syntheses of aldehyde PFBHA O-oximes. Furthermore, there was no RH, temperature, and intermittent

sampling dependence of vapor phase recoveries of the aldehydes, formaldehyde, n-valeraldehyde, and acrolein in terms of the expected PFBHA O-oxime formed.

The sampling tube was connected to the gas bag by Teflon tubing using Tygon collars and butt-to-butt joints. Sampling occurred at specific pump flow rates for known durations corresponding to TLV-TWA or Ceiling Limit exposures as appropriate (Table 3). The sorbent was desorbed with 3-ml hexane by 2-min manual agitation at 25 °C. Some experiments featured different sorbent weights and backup sections, and in certain cases more than triplicate sampling was utilized. This procedure allows a complete mass balance to calibrate the static sampling method[24].

Capacity Testing. Chloroacetone was selected as one representative ketone as it had the lowest ACGIH sample loading (Ceiling Limit of 1 ppm), and it was not sterically hindered[28] . Specific concentrations were generated in 10-L Tedlar gas bags at 1% RH (Tables 4-6). Sampling occurred at average flow rates of 48.90 ± 0.64 mL/min, 9.83 ± 0.66 mL/min, and 2.04 ± 0.12 mL/min at different gas bag concentrations and sampling times. Thus the 48.9 mL/min data were obtained from chloroacetone concentrations of 1.0, 5.0, and 10 ppm sampled at times ranging from 15 min to 315 min. The 9.83 mL/min data were from chloroacetone concentrations of 5, 50, 100, and 250 ppm sampled at times ranging from 80 to 160 min. The sorbent was then desorbed by 3 mL hexane with agitation for 2 min at 25 °C for GC analysis. A similar protocol to that used for chloroacetone was also employed to evaluate acetone,

the ketone with the highest TLV-TWA and which is also not sterically hindered. The selection of the extremes of the capacity spectrum as a screening tool for all aldehydes has been discussed previously[26].

Statistics

All internal statistical comparisons were subjected to analysis of variance types I and II, and to detect significant differences at $p \leq 0.05$ and significant statistical interactions, as well as Student *t* tests for differences of means[29].

RESULTS AND DISCUSSION

Table 1 shows the yields for O-oxime syntheses, corrected for GC/MS TIC purities. All yields are greater than 97.2%, based on 1:1 stoichiometry. Since the yields for methyl *iso*propyl ketone and acetone were also acceptable, and those for di*iso*propyl ketone and for 2,4-hexanedione were not[28], the wet chemical syntheses were of high yield except when the alkyl parts of the ketone were substituted at both β-carbons from the carbonyl carbon.

GC/MS TIC analysis shows the *E*- and *Z*- isomers of the PFBHA O-oximes to have the same molecular ion cluster and fragmentation pattern[22], with a dominant m/z 181 base peak, the 2,3,4,5,6-pentafluorotropylium ion. Average linear dynamic ranges by GC/MS SIM were generally 4.5-20 ng, excepting 0.10-10 ng for chloroacetone (no

E- and *Z*- isomers) and 2-chloroacetophenone (Table 1). All these data are reported for the first time, except for methyl amyl ketone, acetophenone, 2-methylcyclohexanone, and acetone [28]. Generally GC/ECD linear dynamic ranges (not shown here) began at lower injected masses than 1 ng.

Table 2 shows the results of reaction efficiency/O-oxime recovery for wet spiking of ketones. All exceed 87% except for acetone. Acetone spiked at double its TLV-TWA sampled at 10 mL/min 8-hr equivalent was recovered at only 42%, but spiking equivalent to 200 ppm under the same conditions gave 73% recovery. This suggested that the capacity of the sorbent for quantitative sampling was limited to below 40 μmoles but was definitely quantitative at 5 μmoles or below. The theoretical capacity is 160 μmoles. These results agreed with those obtained previously with aldehydes[24-27], and showed that wet spiking the coated tubes for ketones of TLV equivalent of 200 ppm or less would produce quantitative recoveries of PFBHA O-oximes for quantitation purposes, a vital aspect when no commercial standards are available for these O-oxime derivatives.

Initial vapor sampling over 1 hour at TLV-TWA equivalent conditions (that is, using concentrations of 8 x TLV-TWA) showed recovery problems for all ketones at 50 mL/min and 10 mL/min, except for cyclohexanone and methyl butyl ketone at 10 mL/min (Table 3). Since these ketones have 1998 TLV-TWA of 25 ppm and 5 ppm

and are not hindered sterically, the aldehyde technique at 10 mL/min can also be used to sample these two ketones in the 8-hour TWA mode, as well as chloroacetone under Ceiling Limit conditions at 50 mL/min. The aldehyde technique was acceptable for valeraldehyde (TLV of 50 ppm) but was best at 10 mL/min, since breakthrough occurred at 50 mL/min[24]. Thus because most of the ketones had TLV-TWA values >25 ppm, the rest of the ketone vapor evaluations were done at about a flow rate of 10 mL/min, at the TLV sampled for 1 hour. Methyl ethyl ketone, methyl propyl ketone, and 2-methylcyclohexanone sampled under STEL conditions will be successfully sampled with the aldehyde vapor sampling technique.

Vapor sampling efficiencies for ketones at 10 mL/min flow rate for 1 hour in Table 3 exceeded 79% except for acetone, acetophenone, 2-chloroacetophenone, and ethyl amyl ketone where efficiencies varied between 27-36 %. The low recoveries for acetophenone (TLV 10 ppm) and 2-chloroacetophenone (TLV 0.05 ppm) cannot be due to capacity since their TLVs were low. Instead, an inhibition of the gas phase/solid phase reaction relative to the efficient liquid phase/solid phase and liquid phase reactions is implied, probably due to the large flat benzene ring, and restriction of access to the carbonyl group by the other alkyl group. As Table 2 implies, the wet spiking result for acetone was related to capacity, but the gas phase/solid reaction is only about 46% as efficient as the liquid/solid reaction since the acetone loading is about the same in Tables 2 and 3 experiments. Thus the solid/vapor chemisorptive

process differs from the wet chemical process. The result for ethyl amyl ketone is probably of similar origin.

Ethyl and methyl ketones with alkyl straight chain groups from C_1 through C_5 were efficiently sampled for 60 min at their TLV concentrations, as were methyl ketones with isopropyl- and isobutyl- groups and also di propyl ketone. The exception was the *n*-amyl alkyl group in ethyl amyl ketone where the yield was 30% at 200 ppm equivalent. Capacity cannot be the sole reason since 199 ppm equivalent diethyl ketone and methyl isopropyl-, ethyl-, and propyl- ketones between 197-200 ppm equivalent produced acceptable recoveries at the same sampling conditions. Probably, the β carbons of both chains are partially shielded in the gas phase/solid phase reaction though not in the liquid/solid wet spiking experiment. This implies that access of the carbonyl group to the hydroxylamine group of physically adsorbed PFBHA on the solid sorbent is limited for the gas/solid chemisorption of ethyl amyl ketone but not for methyl amyl ketone. Diisobutyl ketone and 2,4-hexanedione show low efficiencies on reaction with PFBHA in their wet chemistry[28].

Previous work showed that the absolute recovery for *n*-valeraldehyde vapor in the dynamic method varied with flow rate, 10 mL/min being better (efficiency of about 100%) than 50 mL/min (efficiency 71-85%)[24-26] . The *n*-valeraldehyde capacity in the gas phase/solid phase[24] was at least 13 μmoles, compared with a maximum of 5

μmoles for both cyclohexanone and methyl propyl ketone for quantitative recoveries in the present vapor phase study.

These results prompted an in-depth examination of the behavior of two unhindered ketones, acetone and chloroacetone, relative to concentration, flow rate, capacity, breakthrough, and sampler parameters.

A 200-mg front section/100-mg back section sampling configuration for 20% PFBHA sorbent showed 31.20±0.44% overall recovery with 26-33% breakthrough after spiking 3 μL acetone (41 μmoles) and then drawing clean air through at 10 mL/min for 2 hr. The same configuration used to sample a 736 ppm gas bag for 2 hr at 9.74 mL/min showed an overall recovery of 49.6±2.7% with breakthroughs of 31-37%. This proved a capacity problem existed for acetone. When 300-mg sorbent coated with 90% PFBHA was spiked with 21.7 μL acetone, the recovery was 58.9±2.9%, an improvement relative to the corresponding 20% coated 200-mg sorbent data of 42.1±6.8% in Table 2. When the 90% sorbent was placed in a 300-mg front section/100-mg backup section configuration, spiked with 21.7 μL again, and clean air then drawn through at 10 mL/min for 2 hr, the overall efficiency decreased to 7.6±1.1% with 10-42% breakthrough. The latter configuration was then used to sample 6009, 3000, and 755 ppm at 10 mL/min for 2-hr periods. The respective overall efficiencies/breakthroughs were: 2.80±0.35%/22-32%; 3.21±0.25%/

25-58%; and 14.1±1.2%/23-28%. Thus increasing the PFBHA content did not solve the capacity problem for acetone, and neither did including a backup section since breakthrough also occurred through the latter. There appeared to be a complex capacity, flow dependence, challenge concentration, and PFBHA coating interactive dependence. The capacity and challenge concentration factors were minimized by using chloroacetone since it had only a Ceiling value, and that concentration was low.

The chloroacetone results for efficiency E in % from gas bag experiments are presented in Tables 4 through 6 for three different average flow rates (F), 48.9, 9.83, and 2.04 mL/min, respectively at different ppm-min exposures P and PFBHA/ketone molar ratios (R). Assuming that the initial chemisorption adsorption isotherm can be described by a Henry (linear) type law indicative of strong adsorption, the best linear regression relationships for 48.9 mL/min in the R range 220 to 25 at $p < 0.05$ for $n=10$ were (Table 4):

$$\text{Log } E = 0.227 \log R + 1.42 \qquad r = 0.9791 \qquad (1)$$

$$\text{Log } E = -0.228 \log P + 2.53 \qquad r = -0.9785 \qquad (2)$$

$$\text{Log } R = -1.0048 \log P + 4.91 \qquad r = -1.000 \qquad (3)$$

Those for 9.83 mL/min in the R range 982 to 10 at $p < 0.05$ for $n=6$ were (Table 5):

$$\text{Log } E = 0.205 \log R + 1.50 \qquad r = 0.7046 \qquad (4)$$

$$E = -0.0020\,P + 108 \qquad r = -0.9160 \qquad (5)$$

$$\text{Log } R = -1.003 \log P + 5.61 \qquad r = -0.9997 \qquad (6)$$

Equation (4) was not significant at $p < 0.05$ but was the best correlation, and is included to compare with equations (1) and (7) for 48.9 and 2.04 mL/min, respectively.

Those for 2.04 mL/min in the R range 25 to 1 at $p < 0.05$ for $n = 5$ were (Table 6):

$$\text{Log } E = 0.735 \log R + 1.07 \qquad r = 0.9796 \qquad (7)$$

$$\text{Log } E = -0.729 \log P + 5.65 \qquad r = -0.9801 \qquad (8)$$

$$\text{Log } R = -0.992 \log P + 6.24 \qquad r = -0.9996 \qquad (9)$$

The log/log relationship was consistently the most statistically significant for all F, except for the linear E versus P relationship for the 9.83 mL/min flow rate.

If 75% recovery is taken as the critical E[30], the critical P/R are 746/101:1 (from equations (2) and (1) respectively), 15577/26:1 (interpolated), and 200,000/10:1 (interpolated) for 48.9, 9.83, and 2.04 mL/min, respectively. The interpolated data

were used because the actual data were close to the critical point, and equation (4) was not significant at $p < 0.05$. Regression analysis showed that P or log P versus F, or P or log P versus R were not linear, as also were log R versus F and log R versus log F.

The only linear relationships at $p < 0.05$ for 200 mg of 20% PFBHA coated sorbent at 75% efficiency were:

$$R = 1.93\,F + 6.48 \qquad r = 1.000 \qquad (10)$$

$$\text{Log } P = -1.76 \log F + 5.88 \qquad r = -0.9990 \qquad (11)$$

$$\text{Log } P = -2.41 \log R + 7.67 \qquad r = -0.9988 \qquad (12)$$

All intercepts are non-zero at $p < 0.05$. Equation (10) shows that R and F are linearly related. At F= 0, R = 6.48. In other work on *n*-valeraldehyde, R was 10.8, and log R versus F was linear, but with a very shallow slope[25]. For *n*-valeraldehyde, R did not vary much, being 12 at 10 mL/min and 17 at 50 mL/min compared with 26 and 103 for these respective flow rates for chloroacetone from equation (10). If chloroacetone is representative of unhindered ketones as is *n*-valeraldehyde for aldehydes, ketones require a much greater excess of PFBHA for quantitative reaction at the same flow rates than do aldehydes. However it is clear

that the initial surface events do not have 1:1 stoichiometry but require at least 12-17 fold excess PFBHA molecules for aldehydes, and 26-100 excess for chloroacetone.

The less bulky carbonyl H of aldehydes allows much better access of the carbonyl group to the hydroxylamine group of surface-adsorbed PFBHA than does chloroacetone to form the initial activated tetrahedral intermediate. The intermediate can still be formed if there are enough excess surface PFBHA molecules at a given flow rate, or if ketone contact time is made longer by lowering F. However, the lower the flow rate, the higher must the sensitivity be of the analytical chemical method. The Tenax TA surface must also play a role in the process since merely increasing PFBHA relative to Tenax TA does not lead to increased efficiency as illustrated by the acetone experiments when R was increased. The benzene ring in the acetophenone molecules, and functional groups substituted at both β carbons to the carbonyl group probably also do not allow ready access of the carbonyl group to the hydroxylamine group of the surface-bound PFBHA.

The extension of the previous aldehyde results to the related carbonyl compounds, the ketones, is not only important to be able to anticipate positive interferences to aldehyde analysis, but also for establishing an analytical method for ketones where the aldehydes may also be positive interferences. Not only is PFBHA a more potent reagent than DNPH since PFBHA reacts quantitatively with conjugated aldehydes

and spiking data generally better match vapor spiking data, PFBHA-based analysis by GC/MS and GC/ECD is better placed for sensitivity and selectivity in a shorter analysis time than the HPLC analysis necessary for the DNPH derivatives. While the PFBHA method has a lower ketone capacity than does activated charcoal (typically >163 μmoles [10]), the PFBHA method can still be used to sample ketones when capacity and steric effects are not problems and when more selectivity is required than in charcoal tube analysis.

CONCLUSIONS

The PFBHA aldehyde dynamic sampling technique was shown to sample ketone vapors also, so that ketones are positive interferences whose presence must be accounted for by the appropriate modification of chromatographic conditions during the chemical analysis step. The best recoveries for ketones occurred at 10 mL/min sampling pump flow rate rather than at 50 mL/min, but especially for ketones whose recommended TLV-TWAs exceeded 25 ppm but not above 200 ppm. Sampling flow rates at 10 mL/min will allow quantitative recoveries for all ketones at TLV-TWA conditions at 200 ppm for 1 hr (200 ppm-hr). Acetone sampling about its TLV or higher under TWA conditions caused breakthrough, while the recoveries of acetophenone, *o*-chloroacetophenone, and ethyl amyl ketone at 200 ppm-hr were affected by steric factors and did not exceed 75 % sampling efficiency. To sample

both aldehydes and ketones together at their TLV concentrations is best done at 10 mL/min flow rate.

ACKNOWLEDGMENTS

This work was made possible through the award of NIOSH Grant RO1 OH03120, and through the financial support of the UCLA Center for Occupational and Environmental Health.

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Table 1. Ketone PFBHA O-Oxime GC/MS Yields for Triplicate Syntheses, and SIM Linear Dynamic Ranges

Ketone	Yield[a] (%)	Oxime Linear Dynamic Range (ng in 2-μL injection)
Chloroacetone	98.4 ± 1.5	0.10 - 10
Diethyl ketone	98.91 ± 0.17	4.5 - 18
Methyl *iso*propyl ketone	98.33 ± 0.36	5.0 - 20
Methyl ethyl ketone	97.8 ± 1.4	5.0 - 20
Methyl propyl ketone	98.1 ± 1.3	5.0 - 20
Methyl *n*-amyl ketone	98.7 ± 1.1	5.0 - 20
Cyclohexanone	98.4 ± 1.6	5.0 - 20
Methyl *n*-butyl ketone	98.67 ± 0.14	5.0 - 20
Acetophenone	98.1 ± 1.9	4.5 - 18
Methyl *iso*butyl ketone	99.2 ± 1.0	4.5 - 18
Ethyl amyl ketone	99.58 ± 0.55	4.5 - 18
2-Chloroacetophenone	97.2 ± 1.4	0.10 - 10
2-Methylcyclohexanone	99.63 ± 0.16	5.0 - 20
Ethyl butyl ketone	98.3 ± 2.8	4.5 - 20
Dipropyl ketone	98.71 ± 0.67	5.0 - 20
Diacetone alcohol	99.56 ± 0.98	5.0 - 20

[a], Corrected for GC/MS purity

Table 2. Desorption Efficiency after Methanol Spikes of the Test Ketones in Triplicate

Ketone (1996 TLV-TWA in ppm, STEL in ppm)	Spiked Ketone (μmol)	Efficiency (%)
Chloroacetone (1 C)	0.031	96.1 ± 5.8
Diethyl ketone (200,-)	4.89	91.1 ± 3.9
Methyl *iso*propyl ketone (200,-)	4.94	92.8 ± 5.6
Methyl ethyl ketone (200,300)	4.50	90.0 ± 4.0
Methyl propyl ketone (200,250)	4.94	92.5 ± 2.6
Methyl amyl ketone (50,-)	1.23	93.3 ± 2.7
Cyclohexanone (25,-)	4.90	87.2 ± 7.0
Methyl butyl ketone (5,-)	0.99	107.00 ± 0.14
Acetophenone (10,-)	2.00	91.5 ± 9.1
Methyl *iso*butyl ketone (50,-)	1.24	98.2 ± 3.5
Ethyl amyl ketone (25,-)	4.89	98.2 ± 2.4
2-Chloroacetophenone (0.05,-)	0.011	95.4 ± 2.7
2-Methylcyclohexanone (50,75)	1.23	99.7 ± 2.3
Ethyl butyl ketone (50,-)	1.23	98.0 ± 3.2
Dipropyl ketone (25,-)	1.23	95.5 ± 6.5
Diacetone alcohol (50,-)	1.20	101.0 ± 4.5
Acetone (750,1000)[a]	294[b]	42.1± 6.8
	39[c]	72.6± 3.4

C, Ceiling (15-min)

[a], 1999 TLV-TWA is 500 ppm and STEL is 750 ppm

[b], Equivalent to sampling double the TLV for 8 hr at 10 mL/min

[c], Equivalent to sampling the TLV for 2 hr at 10 mL/min

Table 3. Sampling Efficiencies for Test Ketone Vapors in Triplicate at Specific Flow Rates, Sampling Times, and Concentrations

Ketone	Gas Bag Concentration (ppm)	Pump Flow Rate (mL/min)	Sampling Time (min)	Sampling Efficiency (%)
Chloroacetone	1.00	49.4 ± 0.64	15	104.7 ± 2.2
Diethyl ketone	199	9.90 ± 0.47	60	95.6 ± 8.5
Methyl *iso*propyl ketone	200	10.01 ± 0.51	60	78.9 ± 4.8
Methyl ethyl ketone	197	9.71 ± 0.43	60	100.1 ± 6.7
Methyl propyl ketone	198	9.6 ± 1.2	60	83.8 ± 4.3
Methyl amyl ketone	49.6	10.08 ± 0.54	60	93.5 ± 4.5
Cyclohexanone	198	9.98 ± 0.11	60	95.5 ± 0.11
Methyl butyl ketone	42.2	9.87 ± 0.22	60	83.8 ± 4.1
Acetophenone	78.4	9.67 ± 0.36	60	26.7 ± 4.8
	9.41	10.10 ± 0.57	60	36.3 ± 2.7
Methyl *iso*butyl ketone	48.5	10.11 ± 0.10	60	96.2 ± 4.4
Ethyl amyl ketone	200	10.11 ± 0.17	60	30.4 ± 2.8
2-Chloroacetophenone	0.44	9.94 ± 0.30	60	36.3 ± 2.7
o-Methylcyclohexanone	53.7	10.37 ± 0.71	60	84.9 ± 3.6
Ethyl butyl ketone	50.8	9.94 ± 0.20	60	97.9 ± 3.2
Dipropyl ketone	49.0	9.81 ± 0.24	60	99.5 ± 4.1
Diacetone alcohol	50.6	9.93 ± 0.24	60	86.2 ± 2.4
Acetone	736	9.740 ± 0.044	120	33.2± 2.4

Table 4. Sampler Capacity Test for Chloroacetone at a Flow Rate of 48.90 ± 0.64 mL/min in Triplicate Experiments

Molar Ratio (PFBHA: Chloroacetone)	Concentration × Sampling time (ppm×min)	Sampling Efficiency (%)
5280:1	15	109.2 ± 2.3
1780:1	45	97.7 ± 5.8
1320:1	60	98.8 ± 5.8
681:1	120	81.2 ± 3.1
539:1	150	90.5 ± 8.9
443:1	180	92.9 ± 6.4
385:1	210	100.7 ± 5.7
307:1	264	103.5 ± 3.0
292:1	276	101.8 ± 3.7
261:1	304	87.4 ± 5.4
220:1	365	88.2 ± 2.9
212:1	376	89.3 ± 3.1
154:1	518	81.8 ± 5.2
136:1	583	81.3 ± 1.6
126:1	634	79.7 ± 3.5
112:1	708	71.6 ± 1.8
102:1	778	72.2 ± 3.2
77:1	1046	71.6 ± 4.4
50:1	1585	60.7 ± 4.9
25:1	3158	55.6 ± 4.0

Table 5. Sampler Capacity for Chloroacetone Vapor at a Flow Rate of 9.83 ± 0.66 mL/min in Triplicate Experiments

Molar Ratio (PFBHA: Chloroacetone)	Concentration× Sampling time (ppm×min)	Efficiency (%)
982:1	405	105.0 ± 9.7
52:1	7627	94.9 ± 9.4
33:1	13156	95.1 ± 4.3
26:1	15577	75.2 ± 1.2
15:1	26100	35.2 ± 4.5
10:1	39000	40.3 ± 7.7

Table 6. Sampler Capacity for Chloroacetone at Flow Rate 2.04 ± 0.12 mL/min in Triplicate Experiments

Molar Ratio (PFBHA: Chloroacetone)	Concentration× Sampling time (ppm×min)	Efficiency (%)
25:1	77844	97.8 ± 5.3
14:1	128478	83.1 ± 5.6
10:1	196250	77.9 ± 5.1
5:1	378500	44.0 ± 9.4
1:1	1950000	10.2 ± 1.3

APPENDIX 5

SYNTHESIS AND HPLC/ULTRAVIOLET DETECTION

ANALYSIS OF THE

O- (2,3,4,5,6-PENTAFLUOROBENZYL)HYDROXYLAMINE

OXIMES OF SELECTED CARBONYL COMPOUNDS

Karina Wiesenthal, Alexander Jehlar and Shane S. Que Hee*

Department of Environmental Health Sciences

and UCLA Center for Occupational and

Environmental Health

10833 Le Conte Avenue

Los Angeles CA 90095-1772

* Author for correspondence

ABSTRACT

The aims were to develop a high performance liquid chromatographic (LC) method with ultraviolet detection (UVD) for O-oximes of O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) of common aldehydes and ketones, and to define the steric limits of the synthetic reaction used to make the PFBHA O-oxime standards for the gas chromatographic (GC) and LC methods. Ten new O-oximes were synthesized with the new optimized method, and their purities demonstrated by GC/electron capture detection (ECD), GC/mass spectrometry (MS), ultraviolet spectroscopy, infrared spectroscopy, and proton and ^{13}C-nuclear magnetic resonance spectroscopy. Ketones substituted at both β-carbons to the carbon carbonyl like diisobutyl ketone and 2,4-hexanedione showed lower synthetic yields by wet chemistry methods. A new C_{18} reverse phase LC/UVD method at 200 nm using acetonitrile/water in both isocratic and gradient elution modes was then developed to resolve sensitively a mixture of 13 pure PFBHA O-oximes. The detection limit was near 100 ng O-oxime/mL or about 14-50 ng aldehyde/mL and the least quantifiable limits were near 500 ng/mL or about 70-250 ng aldehyde/mL, with lower limits for glyoxal, methyl glyoxal, benzaldehyde and acetophenone. Carbonyl compounds in 500 mL water samples were then analyzed in distilled water and tap water by gradient elution. Vapors of *n*-valeraldehyde and acrolein generated in gas bags at concentrations near occupational guidelines were also sampled, desorbed, and then analyzed by either isocratic or gradient elution methods at 200 nm or 254 nm within 30-45 min.

The major analytical method for aldehydes and ketones is high performance liquid chromatography (HPLC)/ultraviolet detection (UVD) of 2,4-dinitrophenyl hydrazones formed by the reaction between 2,4-dinitrophenylhydrazine (DNPH) and the carbonyl group of aldehydes and ketones. The technique has been used for analyses of aldehydes and ketones in air (1-3), water (4), urine (5,6), breath (7), blood (8,9), tobacco smoke (10), food (11), silage (12), and cosmetics (13). Only the formaldehyde DNPH derivative has been utilized for gas chromatography (GC) (14,15) because most of the other 2,4-dinitrophenylhydrazones decompose on-column at high temperatures. DNPH does not react quantitatively with conjugated aliphatic carbonyl compounds (16), and may show variable recoveries on reaction with less reactive carbonyl compounds and at different pH values (17). Some DNPH derivatives are light sensitive (16). The reaction is affected by ozone (18). Efforts to have other analytical methods have resulted.

The major alternative reagent is O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA). Its applications in water and biological analyses have been reviewed (19). PFBHA has been also used in air sampling methods (20,21). The O-oximes of PFBHA are analyzable by GC/electron capture detection (ECD) and GC/mass spectrometry (MS) at pg sensitivity (19). The less sensitive HPLC/UVD method for DNPH can compete only by analysis of a large original sample, injecting a larger volume on-column at the expense of resolution, or total sample desorption/injection at the expense of precision assessment through multiple injections.

However, there are many instances when HPLC analysis of lower molecular weight carbonyl compounds might be favored: analysis of workplace airborne aldehydes and ketones near the

high hygienic air thresholds when the exposing aldehyde is known to be present (22); analysis of ozonolysis disinfection byproducts in water samples (23); and analysis of carbonyl compounds in aqueous biological samples, in foods, in commercial aqueous solutions used as preservatives, embalming fluids, cleaners, and disinfectants; and in taste/odor research (19). There is also a need to have complementary methods to the GC methods with the same standards if resolution, nonvolatility, or GC interferences are problems. HPLC/UVD methods are also relatively inexpensive compared with GC/ECD, but particularly GC/MS. Most laboratories in underdeveloped countries have basic GC/flame ionization detector (FID) and HPLC/UVD capabilities, even though they may not be able to afford GC/MS or even GC/ECD. GC/MS operators also need special expertise to run their instruments effectively.

The aims of the present study were to develop a sensitive and selective HPLC method for typical low molecular weight aldehydes and ketones to define the LC/UVD conditions of use, and to assess the generality of the synthetic reaction to make the essential PFBHA O-oxime derivatives for both the GC and HPLC methods. The most demanding situations relative to sensitivity for the analysis of an environmental sample was adjudged to be ozonolysis byproducts in tap water and analysis of distilled water, since ng/mL aldehyde concentrations are involved. The least demanding environmental sample relative to sensitivity was selected to be samplers that collected samples of known air concentrations of aldehyde vapors. Selectivity requirements re matrix effects are also minimal for these samples. This work is the first report of PFBHA O-oximes being analyzed by a HPLC technique, and allows parallel quantitation to GC-based methods with the same standards if acetonitrile is used as the solvent.

EXPERIMENTAL

Chemicals

The following came from Aldrich, Milwaukee WI: acetaldehyde (99.5%), acetone (99.5%), acetophenone (99%), acrolein (90%), benzaldehyde (99.5%), *n*-butyraldehyde (99.5%), *n*-crotonaldehyde (99%), decafluorobiphenyl (99%), *n*-decyl aldehyde or *n*-decanal (95%), formaldehyde as 37% formalin, glyoxal as 40% aqueous solution, *n*-heptaldehyde or *n*-heptanal (95%), 2-heptanone (99%), pentafluorobenzaldehyde (98%), pentafluorobenzyl alcohol (98%), pyruvic aldehyde (methyl glyoxal) as a 40% aqueous solution, and *n*-valeraldehyde (97%). Fisher Scientific, Tustin, CA supplied Optima grades acetonitrile and hexanes, and carbon tetrachloride. Isobutyraldehyde (99%), 2,6-dimethyl-4-heptanone or diisobutyl ketone (99%), 2-methylcyclohexanone (99%), and 2,4-hexanedione (99%) were from Kodak, Rochester, New York. Deuterated chloroform was from MSD Isotopes, St. Louis MO. PFBHA in the hydrochloride salt form was from Lancaster Laboratories, Lancaster, PA.

Acetonitrile HPLC solvent was filtered through a 0.45 μm Teflon filter (MSI, Westboro, MA). Distilled water was passed through a Millipore Super-Q water deionizing filter system (Millipore, Marlborough, MA), and further filtered through a 0.45 μm nylon filter (MSI, Westboro, MA). ASTM II grade Millipore Super-Q deionized water was produced after further distillation from overnight-refluxed potassium permanganate at pH 2. Helium (99.999%) from Alphagaz, Walnut Creek, CA degassed the HPLC solvents, and 5% methane/argon and nitrogen of the same purity were also from that source. Tenax TA (80/100 mesh) for air sampling came from Alltech Associates, Deerfield, Ill.

Equipment

A Hewlett Packard 1090 Series L liquid chromatograph (Hewlett Packard, Palo Alto CA) preceded a Hewlett Packard 1050 variable wavelength diode array detector with output displayed by a Hewlett Packard 3396 controller/integrator. A Rheodyne 7012 manual injector with a 10-µL injection loop was loaded with a 100-µL injection of sample. The 250-mm x 4-mm stainless steel column contained BioSil ODS-5S C_{18}-reverse phase of film thickness 5 µm.

The gas chromatograph/electron capture detector (GC/ECD) was a Hewlett Packard 5890 with a splitless 30-m x 0.25-mm DB-1701 (1 µm film) chemically bonded fused silica capillary column (J & W Scientific, Folsom, CA) with a constant-current pulse modulated ^{63}Ni-ECD, whose signal was displayed on a Hewlett Packard 3396 controller/integrator. The temperature of the injector and detector was 250 °C. The column temperature program was 50 °C for 1 minute, 50 °C to 250 °C at 5 °C/min, and holding at 250 °C for 4 min. The total run time was 45 min, with an additional 10 min required for thermal reequilibrium to 50 °C. The column flow of 5% methane/argon carrier was 2.5±0.2 mL/min. The flows for the septum purge, make up gas, and anode purge were 2.5 ±0.2, 40±3, and 4.0±0.3 mL/min, respectively.

The same gas chromatograph and column were also interfaced with a Hewlett Packard 5988A mass spectrometer instead of the ECD. The mass spectrometer was a quadrupole with electron multiplier detector over the m/z range 50 to 500 for total ion current (TIC) analyses, and at m/z 181 for selected ion monitoring (SIM) analyses. The temperature of the injector, transfer line, and 70 eV electron impact ion source was 250 °C. The column temperature program was the

same as for GC/ECD. The flow of helium carrier was 3.0±0.3 mL/min. The purge delay was 1 min. 2-μL injections were introduced into the GC/ECD and GC/MS by a 10-μL syringe.

The ^{13}C- and ^{1}H- nuclear magnetic resonance (NMR) measurements were done at room temperature on a Bruker AF200/WB Fourier transform spectrometer (Bruker Instruments, Billerica MA) at spectral widths of 25,000 and 10,000 Hz, respectively. A frequency of 17 Hz was used to decouple the ^{13}C-NMR resonances with a relaxation delay of 20 s. Solutions at >10 mM in deuterated chloroform were contained in 5-mm unfrosted quartz NMR tubes (Aldrich, Milwaukee MI). Tetramethyl silane (TMS) at 1% concentration was the internal reference compound. All spectra were corrected for the method blank.

Infrared (IR) spectra were obtained on a Perkin-Elmer 710B single beam spectrophotometer (Perkin-Elmer, Norwalk CT) using sodium chloride 1-mm cells for liquids with O-oximes dissolved in carbon tetrachloride. Scanning was from 600-4000 cm^{-1} at 4-cm^{-1} resolution.

Ultraviolet spectra were done by a Hewlett Packard 8415A diode array single beam spectrophotometer at 2-mm bandwidth from 190 nm to 820 nm. O-oximes were dissolved in acetonitrile in 1-cm Suprasil cells. Beer's Law plots were done at wavelength maxima and at 254 nm.

General laboratory glassware was from Fisher Scientific. All glassware including solvent reservoirs were first scrubbed in detergent and water, rinsed, soaked in 10% nitric acid overnight,

rinsed again in distilled water, and dried in a dustless oven.

Tedlar gas bags (SKC, Eighty Four, PA) were used to contain static air samples. A 10 microliter syringe (Fisher Scientific) was used to inject liquid carbonyl compounds, and gas-tight syringes were used to transfer concentrated vapors. Compressed air (99.999% purity from Alphagaz, Walnut Creek, CA) was also used. Personal sampling pumps (Model P30A) to sample the gas bag vapors were from DuPont, Wilmington, DE. 20% PFBHA coated Tenax TA (200 mg) in 7 cm long x 5 mm I.D. pyrex tubes contained between two 5-mm glass wool plugs as described elsewhere (20) were used to sample the dry vapors.

Synthesis and Purity of O-Oximes

The major carbonyl compounds selected for study were those that have been detected in surface and the ozonolysis disinfection byproducts in drinking waters (23), and two common aldehydes monitored in workplace air (*n*-valeraldehyde and acrolein) (20), as well as selected industrial chemicals (20,25). These included the carcinogens formaldehyde, acetaldehyde and crotonaldehyde, and the mutagens glyoxal and methyl glyoxal.

The PFBHA O-oximes of aldehydes are not commercially available in the United States. Therefore they had to be synthesized. Hayashi Pure Chemical Industries Limited of Japan now apparently sells the PFBHA O-oximes of C1-C8 and C10 linear aldehydes, of C4 and C5 branched chain aldehydes, and of glyoxal and methyl glyoxal. The syntheses and purities for the derivatives of formaldehyde, acetaldehyde, *n*-heptanal, *n*-decanal, and glyoxal are detailed by our

research group elsewhere (24), as are those for acrolein, crotonaldehyde, and *n*-valeraldehyde (20). New derivatives were synthesized for acetone, acetophenone, benzaldehyde, *n*-butanal, and methyl glyoxal that were studied for HPLC purposes. The other carbonyl compounds not studied for HPLC (2-heptanone, isobutyraldehyde, diisobutyl ketone, 2-methylcyclohexanone, and 2,4-hexanedione) were used to assess the influence of steric hindrance on the synthetic reaction. The synthesis of their PFBHA O-oximes are reported also for the first time. The mass spectra of all these O-oxime derivatives are available elsewhere (25). The pure PFBHA O-oximes were used for HPLC and GC standardizations.

The general method of optimal synthesis of about 200 mg of PFBHA O-oxime for both aldehydes and ketones was to use 1.15:1 molar ratios of PFBHA/monocarbonyl compound or from 2.30:1 molar ratios of PFBHA/dicarbonyl compound. The pure PFBHA was dissolved in 10 mL of ASTM II water in a 16-mm x 150-mm Teflon-lined screw-capped Kimax culture tube (Fisher Scientific). The carbonyl compound was dissolved similarly in 2 mL of ASTM II water if water soluble, or of methanolic aqueous solution if not. For the latter, the minimum amount of methanol was added to 2 mL aqueous solution in 10-μL aliquots to promote water solubility on shaking. Five times the methanol added to solubilize the carbonyl compound was then added to the PFBHA solution. The aldehyde was then added dropwise to the PFBHA solution with manual agitation. After adding all the aldehyde solution, the PFBHA/aldehyde mixture was then vortexed vigorously after putting on the screw cap. Any pressure was relieved by loosening the cap. The agitated turbid solution was then immediately microwaved at the lowest power setting on the turntable of a commercial 850-W 2450 MHz microwave oven (Sharp Model R-3A85,

Sharp Electronics Corp., Mahwah NJ) until the first bubble appeared (about 80-85 °C water temperature after about 10 s heating). The tube was allowed to sit in the oven for one minute (cap on) before removing to cool to room temperature. The solution was then immersed in an ice bath for 30 min. If a precipitate or a second layer appeared, the top aqueous layer was removed by Pasteur pipet and transferred into another screw-cap tube for further extraction (three times each with 1 mL of hexane). The combined extracts were added back to the residual solid or liquid from the synthesis. If precipitate or a second layer still remained on vortexing the latter mixture, more hexane was added in 0.5 mL aliquots until dissolution on manual shaking. After centrifugation in a bench centrifuge for 2 min at 900g, the top hexane layer was removed by Pasteur pipet, and any residual liquid washed two more times with 0.5 mL aliquots of hexane. The extracts were combined in a V-vial of appropriate volume, and the hexane evaporated under nitrogen passed through a 20/40 mesh 200/100 mg large charcoal tube (SKC, Eighty Four, PA), until the container was no longer cool. The vial was then placed in a vacuum desiccator that contained indicating Drierite desiccant until constant mass was achieved. The yields were calculated, and a known mass taken for purity estimation through ultraviolet, Fourier-transform infrared, ^{13}C- and ^{1}H-nuclear magnetic spin resonance spectroscopies, and GC/MS. In GC/MS and GC/ECD runs, the purity was calculated by correcting for the content of unreacted PFBHA, pentafluorobenzyl alcohol, pentafluorobenzyl aldehyde, any other peaks that corresponded to other carbonyl compounds, and those that were not from the reagents and solvents (24).

Standard curves of detector response versus mass injected for the GC methods using 5 concentrations of 1-μL hexane or acetonitrile injection volumes were done for each O-oxime in

triplicate (interrun analysis), each tube containing 400 ng/mL of decafluorobiphenyl internal standard (m/z 334 in selected ion monitoring (SIM) GC/MS). One of the triplicate samples was also injected three times to define intrarun precision. All O-oximes were also combined in hexane or acetonitrile. The slopes (response factors) were determined by linear regression after the linear regions were defined. The formaldehyde O-oxime eluted at 4.59 min and the last O-oxime to elute, the derivative for *n*-decanal, eluted at 32.85 min. One GC cycle took 55 min.

Essentially the same procedure using the pure PFBHA O-oximes was performed for HPLC standardizations using acetonitrile solutions, but not using the internal standard. The molar response factors and the relative molar response factors (RRFs) were also determined at the optimized isocratic and gradient elution conditions. This provided absolute PFBHA O-oxime determinations independent of reaction efficiency in the water sample and the factors that affected PFBHA O-oxime recoveries for HPLC quantification.

HPLC Method Quality Control

A known weight of each pure O-oxime (about 7-40 mg) was dissolved by acetonitrile to 10-mL in a volumetric flask to create a known high concentration stock. All solutions were ultrasonicated at 50 °C for 1 min. Subdilutions of O-oximes from 50 μg/mL substock were prepared in 25-mL volumetric flasks to generate five concentrations ranging from 0.1-10 μg/mL for each O-oxime, and for the mixture. Intrarun (triplicate injections of the same solution) and interrun precisions (analysis of the triplicate samples) were calculated. Storage of all samples after use was at -20 °C. The same concentrations are obtained on thawing to room temperature,

and then ultrasonicating at 50 °C for 1 min. This procedure allowed reproducible standard solutions on dilution for at least 6 months.

HPLC Method Development

The next objective after obtaining pure PFBHA derivatives was to select the appropriate LC column. The initial column was a 250 mm x 6.2 mm Supelcosil LC-1 reverse phase column of 5 μm particle size, employed originally to separate toluene, xylenes, and ethyl benzene, all nonpolar aromatic hydrocarbons. Poor resolutions of O-oximes occurred and the long chain oximes never eluted even in 100 % acetonitrile. The Biosil ODS-5S C_{18} reverse phase column had been used to separate benzyl alcohol, benzaldehyde, and the ester, methyl benzoate. These more polar aromatic compounds proved closer in polarity to the O-oximes than were the aromatic hydrocarbons.

The next step after column selection was to evaluate the water miscible organic solvents, acetonitrile, methanol, and tetrahydrofuran in various aqueous solvent compositions (0, 25, 50, 75, and 100 % v/v) under isocratic conditions at different wavelengths, flow rates, and column temperatures. Gradient elution conditions were then optimized using the isocratic run data as guide. The linear dynamic range at the optimum gradient elution and isocratic conditions at 200 nm was then determined for 10-μL loop injections of 0, 0.1, 0.5, 0.75, 1.0, 2.4, 5.0, 7.5, 10, 25, and 50 μg O-oxime/mL acetonitrile solution. Some experiments were also performed at 254 nm for concentrations that were 25 times higher.

Analysis of Los Angeles Tap Water and Millipore Deionized Water

Volumes of 500 mL of Los Angeles tap water and deionized super Milli-Q water samples were analyzed in triplicate with the optimized gradient elution method, in addition to reagent blanks. The 500-mL volume was selected based on the lowest quantifiable level of the least sensitive aldehyde at 200 nm wavelength.

The 500-mL tap water sample had 10.0 mL of 0.1 N sodium thiosulfate added to reduce hypochlorite and ozone residual from ozonation/chlorination disinfection utilized for Los Angeles tapwater. A volume of 50 mL of 10 mg/mL PFBHA in Millipore deionized water was then added. This step was the starting point for the 500-mL deionized water sample. The pH was adjusted to about 2.0 (range 0.9-2.0) with 10% HCl using 25 µL aliquots with stirring. The sample was then subjected to microwaving until the first bubbles appeared. After cooling to room temperature, 5 mL of 18 N sulfuric acid was added with stirring to minimize excess PFBHA. Each sample was then added to a 1-L separatory funnel with Teflon stopcocks and stoppers. Hexane extractions (3 x 10 mL) then ensued. The hexane extracts for each sample were combined into individual 50-mL round bottom flasks. Each hexane solution was evaporated to about 3 mL by rotary evaporation at 45 °C. Each 3-mL sample was transferred to a preweighed V-vial, and the rest of the hexane evaporated to constant residue weight under a gentle stream of nitrogen. After reweighing, the residue was redissolved in 0.600 mL of acetonitrile, constituting a 833.3-fold concentration over the original concentration. Distilled water treated in parallel served as the reagent blank to correct for background contamination. Blank runs for reagents were also done in parallel, as were positive controls containing aldehyde mixtures of known ppb

composition in 500 mL distilled water solution to determine recoveries (10 ng/mL of formaldehyde, 10 ng/mL of acetaldehyde, 3.0 ng/mL of *n*-butanal, 2.0 ng/mL of *n*-heptanal, and 1.0 ng/mL of *n*-decanal, the concentrations detected by Glaze et al (23,26)).

HPLC injection using 50 μL to load the 10-μL injection loop then followed, for isocratic or gradient elution conditions. The optimum isocratic conditions were 57% acetonitrile/43% water at 39 °C and flow rate 0.80 mL/min. The optimum solvent linear gradient relative to acetonitrile and water at 39 °C and 0.80 mL/min was: 47.5% acetonitrile at 0 min; 53% at 16.9 min; 60% at 22.5 min; 65% at 28 min; 73% at 36.70 min; 75% at 38 min; 80% at 45 min; 95% at 60 min; and 100% at 70 min, the latter step to clean the column before recycling. As soon as the last analytical peak had eluted as evidenced by integrator tick marks, the solvent for isocratic and gradient elution runs was set at 100% acetonitrile to shorten analyses and to clean the column, as evidenced by a flat baseline before recycling to the initial conditions.

The PFBHA O-oximes were determined by the method of external standards using the pure PFBHA O-oximes that were produced during their syntheses. Acetonitrile was always the HPLC injection solvent. O-oxime concentrations in ng/mL were converted to carbonyl compound equivalents by multiplying by the molecular weight of the aldehyde divided by the molecular weight of the PFBHA O-oxime. Peaks and quantitative contributions due to the hexane solvent, reagents, and deionized water were assigned and corrected for. Distilled water provided the reagent blank data that were subtracted from tap water data.

Gas Bag Sample Analysis

The static air sample was generated by first half-filling the 10-L gas bag with pure air, injecting the calculated mass of pure valeraldehyde liquid and acrolein in air (20) with a 10-μL syringe and gas-tight syringe, respectively, to be equivalent to the 1997 TLV-TWA (22) over 8 hours of sampling (176 mg/m^3 for valeraldehyde and 0.23 mg/m^3). After adding another 5.0 L of air, the bag was heated with a hot-air hair dryer. After cooling, the vapor was sampled at 10 mL/min using 200 mg solid sorbent Tenax TA (80/100 mesh) coated with 20% PFBHA in a glass tube as described elsewhere (20). The solid sorbent was desorbed with 2 mL hexane in a small centrifuge tube for 2 hours with shaking every half hour as recommended (20). This technique allowed the validated air sampling method to be used without revalidation for another desorbing solvent. The solution was centrifuged for 1 min on a desk centrifuge and the supernatant transferred to a V vial . The hexane solvent was evaporated in a gentle stream of nitrogen using a Pasteur pipet in a fume hood. A volume of 2 mL of acetonitrile was then added to the valeraldehyde tubes and 1 mL to the acrolein tubes. Both sets of solutions were ultrasonicated for 1 min at 50 °C. A volume of 2 μL of the valeraldehyde PFBHA O-oxime solution was made up to 1 mL in acetonitrile in a graduated 1-mL test tube, and HPLC analysis then ensued. The acrolein solution could be injected directly without further dilution. All experiments were done in triplicate.

Results and Discussion

Table 1 shows the yields for all the PFBHA O-oximes of the HPLC study, some physical characteristics of the parent carbonyl compounds, the PFBHA O-oxime retention times for the

optimized isocratic and gradient elution HPLC and GC/ECD methods, the molar relative response factors RRF at 200 nm relative to the O-oxime of benzaldehyde as 100%, the intrarun and interrun precisions at 100 ng O-oxime/mL for the optimized gradient elution HPLC method, and the equivalent concentrations of carbonyl compounds in one sample of Los Angeles tap water quantified with the optimized method for distilled water.

Synthetic yields of O-oximes

Most PFBHA O-oxime synthetic products showed *E*- and *Z*- isomers on TIC GC/MS analysis, but all yields include both isomers.

All synthetic yields for compounds in Table 1 exceeded 84%. The yields for the eight already-reported O-oximes (20,24) agreed with previously reported results at $p \leq 0.05$ (Student *t* test). The yields for the five other carbonyl compounds in Table 1 (acetone, acetophenone, benzaldehyde, *n*-butanal, and methyl glyoxal) are reported for the first time.

Table 2 shows the yields for the other carbonyl compounds used to assess steric hindrance dependence of the addition reaction. These yields are also reported for the first time. The lowest yields were for those compounds where steric hindrance was a factor to formation or further reaction of the tetrahedral addition intermediate. Yields decreased for ketones substituted with alkyl branches at both β-carbons to the carbonyl carbon. Thus, diisobutyl ketone had a final yield corrected for purity of below 0.2%; 2,4-hexanedione had a corresponding yield of 48%. The presence of non O-oxime impurities was the major problem for these two ketones as revealed by

GC/MS TIC analyses. Yields for the straight chain and the cyclohexyl ketones exceeded 80%. Thus all aldehydes so far synthesized and ketones that are straight chain that are monocarbonyls or diketones that are not sterically hindered produce yields ≥75%. GC/MS and GC/ECD showed the presence of the *E*- and *Z*-isomers of the O-oximes of asymmetric carbonyl compounds (25), though the optimized gradient elution HPLC method resolved more isomers (Table 1; Figure 1).

In the original synthetic method, the reaction was quenched immediately in an ice bath for 30 min after the microwave step (20,24). This method is still effective for straight chain aliphatic aldehydes and glyoxal, but much lower yields for methyl glyoxal and ketones occur. The extra standing time of 1 minute for the hot solution plus the slow equilibration to room temperature allows the reaction to go to completion for ketones. Incomplete reaction for methyl glyoxal was signalled by the presence of its monosubstituted O-oxime.

Spectroscopic Characterization

Mass Spectra

All O-oximes had m/z 181 as their base peak in their mass spectra, and this peak facilitates sensitive and selective SIM for GC/MS (25). Observed linear ranges were generally between 30 ng-200 ng for TIC analyses, and an order of magnitude lower for SIM analyses. The usual intrarun precision was <10% in the linear range. The usual interrun precision was <17%.

Ultraviolet Spectroscopy

Molar absorptivities (ϵ) at the long wavelength maximum of 264 nm varied between 491-667

L cm^{-1} mol^{-1} for PFBHA O-oximes of aliphatic monocarbonyl aldehydes or a midpoint of 579 L cm^{-1} mol^{-1}. In contrast, benzaldehyde PFBHA O-oxime had an ϵ of 17,260±270 L cm^{-1} mol^{-1}. Methyl glyoxal PFBHA O-oxime, a disubstituted derivative, had an ϵ of 8,410±100 L cm^{-1} mol^{-1}. The ϵ of the only other reported disubstituted PFBHA O-oxime for glyoxal was 13,200 L cm^{-1} mol^{-1} at this wavelength (24). Thus the benzaldehyde, methyl glyoxal, and glyoxal PFBHA O-oximes can be detected, on average, with sensitivities 31, 15, and 24 times, respectively, those of aliphatic monocarbonyl PFBHA O-oximes at this wavelength.

At the short wavelength maximum of 200 nm, the ϵ varied from 10,200-15,000 L cm^{-1} mol^{-1} for the aliphatic monocarbonyl derivatives. The midpoint ϵ for aliphatic monocarbonyl PFBHA O-oximes is thus 12,600 L cm^{-1} mol^{-1}. The corresponding ϵ for benzaldehyde was 26,210±500 L cm^{-1} mol^{-1}, and that for methyl glyoxal was 10,650±580 L cm^{-1} mol^{-1}. The literature value for glyoxal was 13,600 ±540 L cm^{-1} mol^{-1} (24). Thus the benzaldehyde, methyl glyoxal, and glyoxal PFBHA O-oximes can be detected on average with sensitivities 2.1, 0.85, and 1.1 times, respectively, those of aliphatic monocarbonyl PFBHA O-oximes at this wavelength. The ϵ are far more uniform than at 264 nm at least for aliphatic mono- or di-carbonyl compounds, and are also high. For a 1 cm pathlength, an absorbance of 0.0010 is equivalent to 80 nM or about 18 µg/L or 18 ppb of formaldehyde PFBHA O-oxime.

At the second short wavelength maximum of 222 nm, PFBHA O-oximes of aliphatic monocarbonyl derivatives had ϵ varying from 2,970 to 4,480 L cm^{-1} mol^{-1}, or a midpoint of 3,730 L cm^{-1} mol^{-1}. Those for benzaldehyde and methyl glyoxal were 8,140±200 and 13,300±400 L

cm^{-1} mol^{-1}, respectively. The corresponding molar absorptivity for glyoxal was 10,900±780 L cm^{-1} mol^{-1} (24). Thus the benzaldehyde, methyl glyoxal, and glyoxal PFBHA O-oximes can be detected on average with sensitivities 2.2, 3.6, and 2.9 times, respectively, those of aliphatic monocarbonyl PFBHA O-oximes at this wavelength. The aliphatic mono- and di-substituted PFBHA O-oximes can now be distinguished from each other unlike at 200 nm.

In general for the aliphatic monosubstituted PFBHA O-oximes at their three maxima, the ultraviolet sensitivity ratio of the molar absorptivity midpoints for 200, 222, and 264 nm was 22:6.4:1.0, and should be diagnostic for other aliphatic monosubstituted PFBHA O-oximes too. In contrast, this ratio for the benzaldehyde derivative was 1.5:0.47:1.0, that for methyl glyoxal was 1.3:1.6:1.0, and that for glyoxal was 1.0:0.83:1.0. These ratios can be used to confirm the suspected presence of these three derivatives during HPLC runs. The sensitivity at 200 nm relative to at 254 nm, the most common fixed wavelength used in HPLC, was between 30-40 times greater for aliphatic monosubstituted PFBHA O-oximes. Thus 200 nm was the wavelength of analytical choice since it was the most sensitive, and variation in ϵ and hence LC/UVD response factors were expected to be small.

Infrared Spectroscopy

The infrared spectra were consistent with the presence of pure PFBHA O-oximes.

The infrared spectra revealed strong C-O stretching at 1125-1141 cm^{-1}, moderate C-H stretches at 2825-3225 cm^{-1}, moderate C-C aromatic stretches at 1652-1675 cm^{-1}, and weak N-O stretches

at 920-995 cm^{-1}. These observations agree with prior results (24). Carbon tetrachloride C-Cl absorption obscured the C-C stretch at 1500-1510 cm^{-1}, and the weak C=N stretch at 1400-1700 cm^{-1}. GC/FT/IR analysis is therefore recommended at 1125-1141 cm^{-1} in the fingerprint region.

NMR Spectroscopy

Both types of NMR spectra were consistent with the presence of pure PFBHA O-oximes.

The ^{1}H-NMR spectra showed the following respective *E*- and *Z*- isomer splittings downfield of TMS in ppm: oxime protons, 6.47-6.78/7.07-7.82 with respective coupling constant J values of 5.50-7.49/5.99-7.43 Hz; ether protons, 5.14-5.33/5.09-5.27; and the protons on the carbon atom α- to the oxime carbon atom, 2.27-2.41/1.82-2.15 with respective J values of 5.15-5.68/6.57-7.28Hz except for 7.82 ppm for the benzaldehyde and acetophenone derivatives. The resonances of the protons in the methylene groups of long alkyl chains were superimposed between 1.06-1.44 ppm with multiplets. Terminal methyl groups resonated at 0.87-0.88 ppm (triplet). Benzaldehyde and acetophenone aromatic protons on carbons 2 and 6 resonated at 7.29 ppm, on carbons 3 and 5 at 7.41 ppm, and on carbon 5 at 7.57 ppm.

The ^{13}C-NMR spectra showed resonances between 141.31-160.00 ppm for oxime group carbons, and 60.00-65.11 ppm for ether carbons. The aromatic carbons of the fluorinated ring had the following resonances in ppm: 138.71-145.98 for *ortho*-, 142.91-151.89 for *meta*-, and 146.98-154.49 for *para*- positions. The O-oxime of *n*-butanal showed the following respective *E*- and *Z*- resonances in ppm downfield of TMS: the oxime carbon, 152.4/153.5; the α-carbon to

the oxime carbon, 27.52/31.4; and the β-carbon to the oxime carbon, 19.41/19.8. Isobutyraldehyde in contrast showed no E- and Z- isomers. The *E*- and *Z*- resonances for the formaldehyde, acetaldehyde, *n*-heptanal, *n*-decanal, and glyoxal derivatives are provided elsewhere (24). The signals for both aromatic carbonyl and PFBHA aromatic rings (24) were superimposed for *ortho*-, *meta*-, and *para*- carbons. The ring carbon of side chain attachment had resonances at 113.50-114.49 ppm for the fluorinated ring, and 127.2 ppm for the aromatic carbonyl ring. The other aromatic carbonyl ring resonances in ppm were: *ortho*-, 128.76; *meta*-, 131.03, and *para*-, 130.3. The methyl group on methyl glyoxal had an upfield signal of 9.64 ppm because of shielding of the two oxime carbons, compared with 13.66-17.07 ppm for unshielded terminal methyl groups of straight chain compounds, and 19.78 ppm for the two equivalent methyl groups of isobutyraldehyde. The β-carbon in the long chain to the oxime carbon for isobutyraldehyde resonated at 29.35 ppm. Superposition occurred for unshielded methylene carbon atoms of long chain aldehydes between 29.00 to 32.27 ppm. The NMR data were consistent with previously published data on other pure aldehyde PFBHA O-oximes (24).

HPLC Method Development

Isocratic Conditions

At 1 mL/min flow rate at 39 °C, pure methanol could not elute all the long chain O-oximes in Table 1 injected as a 10 μg/mL mixture within one hour, while pure acetonitrile and tetrahydrofuran eluted all. All peaks eluted in 36 min in pure acetonitrile, and there was good peak shape and sensitivity, but poor resolution. Tetrahydrofuran did not provide sufficient resolution for the short chain O-oximes at increasing water content. At 39 °C, water produced

good resolution but poor sensitivity, very broad peaks, and runs took hours. Partial resolution occurred for acetonitrile/water concentrations. Acetonitrile contents of 25% and 50% still had very long runs >120 min at 39 °C. Since partial resolution was achieved at 75%, the optimum acetonitrile composition was between 50 and 75% at 39 °C and 1.0 mL/min.

Complete resolution under isocratic conditions took about 115 min, and was achieved with 57% acetonitrile at 39 °C and flow rate 0.80 mL/min (Table 1). At 1.5 mL/min, acrolein, *n*-crotonaldehyde, and *n*-valeraldehyde were not resolved. At 0.4 mL/min, chromatographic runs were 160 min, and sensitivity decreased due to peak broadening especially for the late eluting compounds. The detection limits for injection loop volumes of 5, 10, and 200 μL were 1 μg/mL, 100 ng/mL, and 10 ng/mL injected O-oxime concentration in acetonitrile at optimal isocratic conditions. The 200 μL injection showed very poor resolution, and the 5 μL injection was not sensitive enough. Thus 10 μL injection loop volume provided the best resolution and sensitivity, and thus was selected for all subsequent work.

The logarithm of the retention time t_R corrected for dead volume time t_M denoted as t_R' plotted against O-oxime molecular weight MW was linear for all monocarbonyl data including the monosubstituted methyl glyoxal at the optimized isocratic conditions.

$$\log t_R' = 0.0091\ MW - 1.153 \qquad r=0.9503 \text{ for } n=9,\ p \leq 0.05 \qquad (1)$$

A similar regression emerged for the same compounds when the capacity factor k' replaced t_R'

(27):

$$\log k' = 0.0093\ MW - 1.021 \qquad r=0.9523 \text{ for } n=9,\ p \leq 0.05 \qquad (2)$$

where

$$k' = (t_R/t_M - 1) = t_R'/t_M \qquad (3)$$

The fully PFBHA substituted dicarbonyl compounds did not obey regression equations (1) or (2).

Equations (1) and (2) allow t_R', t_R, and k' to be determined for any straight chain aldehyde or ketone under the optimum isocratic conditions for PFBHA O-oxime congeners between and including C1 to C10 carbonyl compounds with one carbonyl group. The model does not distinguish isomers, either skeletal or *E*- or *Z*-.

Since the plate count N is dependent on t_R and the peak half-width $W_{0.5}$ through equa 4) (27):

$$N = 5.54\ (t_R/W_{0.5})^2 \qquad (4)$$

Equations (3) and (4) lead to equation (5):

$$\log t_R = 0.5 \log N - 0.3718 + (\log W_{0.5})/2.303 = \log [t_M(k' + 1)] \quad (5)$$

For the acetophenone derivative under isocratic conditions, t_R = 39.785 min (Table 1) and $W_{0.5}$ = 0.746 min so that N is about 15,800. Similarly, the respective N values for the formaldehyde and *n*-decanal derivatives are 12,500 and 17,700. N is not constant because $W_{0.5}$ increases nonlinearly as t_R increases.

Gradient Elution

A complete gradient elution HPLC cycle to resolve all 13 PFBHA O-oximes including column cleaning and recycling to the original conditions took 90 min. A complete GC cycle, in contrast, took 55 min.

Gradient elution provided better peak shapes and resolution, as well as shorter run times than isocratic conditions. The last O-oxime peak for *n*-decanal eluted between 51-53 minutes (Table 1 and Figure 1), compared with 112 min at optimized isocratic conditions. Other peaks not related to PFBHA O-oximes had to be eluted to ensure the column was clean, however, prolonging run times. Acetonitrile gave peaks at 1.7 min (large), 2.4 min (moderate), and 4.7 min (small). Other background peaks occurred at 55.5 min (large) as shown in Figure 1, 66.2 min (large), 72.0 min (moderate), and 77.5 min (small) in the acetonitrile mixture of O-oximes. That better resolution was attained under gradient elution conditions is also attested to by the presence of more resolved *E*- and *Z*- isomers of the O-oximes than at isocratic conditions. Both peaks or one peak can be used for quantitations depending on if there are any interferences. One-peak analysis is less

sensitive than 2-peak analysis. In the blanks without added PFBHA, nonpolar substituents absorbing at 200 nm present in distilled water, tap water, hydrochloric acid, and hexane are coextracted with the PFBHA oximes after PFBHA is added and upon hexane extraction and concentration. Any interfering peak areas from these blanks were subtracted before PFBHA O-oxime concentrations were calculated. Such interferences were minimal for tap water, but more important for distilled water (Figure 1). The corrected distilled water chromatogram is effectively the PFBHA reagent blank for the corrected chromatogram for tap water. Thus further subtraction yielded the results for tap water in Table 1.

In gradient elution, the $W_{0.5}$ in equation (5) is kept as constant as possible through changing the Snyder solvent polarity index (27). Log t_R then varies almost linearly with log N.

Stepwise dilutions showed that at 200 nm the gradient elution method at twice the noise level was at about 100 ng/mL PFBHA O-oxime with intrarun coefficients between 3.0-29% (Table 1). The interrun CV was between 9.0-70% (Table 1). Above this concentration, intrarun CVs decreased, being 2-15% at 0.5-1.0 μg/mL, and 0.1-7.0 % at 2.5-10 μg/mL.Thus, while detection at 100 ng/mL PFBHA oxime is possible (or about a minimum of 14 ng/mL of formaldehyde equivalent), quantitation with acceptable precision occurs >500 ng/mL (about 70 ng/mL of formaldehyde equivalent), and is the general least quantifiable limit (LQL). The linear dynamic range was from 0.100 to 25 μg/mL (about 0.014 to 4 μg/mL formaldehyde equivalent) corresponding to an absorbance range of 0.001 to 0.010.

The HPLC RRFs relative to the benzaldehyde reference of 100% at 200 nm in Table 1 for the optimized gradient elution method show a clear distinction between PFBHA O-oximes of the aromatic carbonyl compounds (96 and 100% for acetophenone and benzaldehyde, respectively) and those of other carbonyl compounds (35% for formaldehyde to 55% for acetone). RRFs increased from formaldehyde through to *n*-heptanal but decreased to *n*-decanal for the long chain aliphatic aldehydes. Table 1 shows that the average RRF was 54±21% of CV 39% for all compounds, with the 11 aliphatic O-oximes as a group being 46±7 %, CV 15%. The latter is consistent with uniform ϵ values at 200 nm for the aliphatic mono- and di-substituted carbonyl compounds.

Tap Water and Deionized Water Analyses

The only solid residue detected after hexane extract evaporation before resolubilization in acetonitrile was 1.8 mg for tap water. Excess PFBHA eluted at 5.9-6.2 min, and was the largest peak (Figure 1). PFBHA was well separated from the closest PFBHA O-oxime peak for formaldehyde. This is an advantage over the GC method where PFBHA elutes between the PFBHA O-oximes of formaldehydes and acetaldehyde, which requires the addition of sulfuric acid before hexane extraction to minimize potential PFBHA interference with these two PFBHA O-oximes. A time of 110 min allowed elution of all peaks. A small peak at t_R=6.9 min was contributed by hexane. Peaks at 3.6 min (small), 10.8 min (small), 39.4 min (moderate), 44.1 min (small), 48.0 min (small), and 49.7 min (moderate) were noncarbonyl peaks contributed by the other reagents except sodium thiosulfate. Small noncarbonyl peaks from sodium thiosulfate were at t_R of 25.8, 36.3, and 38.5 min. The derivatization reaction preferentially selected out

carbonyl compounds, and any hexane co-extracted interferences would have to be nonpolar and of large molecular weight >200 to interfere with PFBHA O-oxime resolution.

As expected, the concentrations of carbonyl compounds in the distilled deionized water uncorrected for presence of reagents were low. The concentrations in ng carbonyl compound/mL were: acetone, 2.5; acetophenone, 1.8; benzaldehyde, 6.8; crotonaldehyde, 0.024; formaldehyde, 0.13; *n*-valeraldehyde, 9.7; and *n*-heptanal, 11. The rest of the aldehydes were below their LQLs.

The carbonyl compound concentrations in tap water were generally higher than in distilled water (Table 1). Crotonaldehyde is probably the most uncertain concentration. The CVs are of the order of 10-17% for the interrun replicates. Of the 36 ng aldehydes/mL detected, 39% was *n*-heptanal, 15% was formaldehyde; 13% was acrolein, 6.9% was *n*-decanal, and 6.4% was *n*-butanal, comprising 80% of the mass of aldehydes analyzed. These aldehydes are the classical ozonolysis byproducts from bacterial precursors (23,26).

Glaze et al (23,26) have found between 3.8-25 ng/mL of formaldehyde, 9.5 ng/mL of acetaldehyde, 3.1 ng/mL of *n*-butanal, 2.0-2.3 ng/mL of *n*-heptanal, and 1.1 ng/mL of *n*-decanal in some finished waters. The recoveries of the PFBHA methods for water carbonyl compounds exceed 80% (23,24,26), and this was also confirmed in the present study by the >80% recoveries of the components of a defined aldehyde mixture based on the above Glaze et al data.

The nineteenth edition of *Standard Methods for the Examination of Water and Wastewater*

contained Method 6252 using PFBHA in a liquid-liquid extraction mode. The latest twentieth edition in 1998 contained a slightly revised method (28). In the latter, ammonium chloride or ammonium sulfate were recommended as reducing agents to ensure hypochlorite ion did not interfere with the PFBHA derivatization reaction. The addition of sodium thiosulfate or potassium iodide was recommended to reduce excess ozone. EPA Method 556 published in 1998 (29) utilized the same methodology except that it warned that sodium thiosulfate could partially reduce unspecified organic material from microorganisms to aldehydes. Both of these standard methods differ from the technique used in the present work by utilizing a potassium phthalate buffer solution for the PFBHA derivatization instead of a pH 2.0 PFBHA solution. Method 6252 utilized a pH 6.0 buffer, whilst the EPA method used a pH 4.0 buffer. There has been no published comparison of the two methods.

Analysis of Desorbed PFBHA O-Oximes of n-Valeraldehyde and Acrolein Vapors

The analysis for desorbed acrolein and *n*-valeraldehyde could be done either isocratically or by gradient elution, but isocratic analysis was much quicker for acrolein and gradient elution faster for n-valeraldehyde (Table 1), with acetonitrile content being set at 100% as soon as the analyte peak eluted. The cycle time for analysis for acrolein was 30 min and that for n-valeraldehyde was 45 min. Both analyte peaks were well separated from the huge PFBHA peak.

A wavelength of 200 nm was satisfactory even for the acrolein PFBHA O-oxime where sensitivity requirements were highest since acrolein has the lowest TLV-TWA of any aldehyde (22). For valeraldehyde, if a wavelength of 254 nm was used at gradient elution conditions, only

a two-fold dilution was necessary for quantitation. The HPLC method has flexibility since the acetonitrile volume added after the hexane evaporation and any subsequent dilution ratio can be varied at will depending on the hygienic guidance value, and in addition 200 nm, 222 nm, 254 nm, and 264 nm can be used as analytical wavelengths as discussed above in the ultraviolet spectroscopy section. Thus HPLC can provide fast, accurate, selective analysis of aldehyde vapors near their hygienic guidance levels, and there is no reason why the technique will not be effective for sterically unhindered ketones also, especially since their TLV-TWA values are larger than aldehydes of the same number of carbon atoms (22).

Acknowledgments

This work was funded by NIOSH Grant RO1 OH03120, the UCLA Center for Occupational and Environmental Health, and the Metropolitan Water District of Southern California Agreement Number 3399. Part of this work was presented at the American Industrial Hygiene Conference and Exhibition, May 17-23, 1997, Dallas, Texas, as Abstract 277.

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Table 1. Carbonyl Compound Analysis by HPLC and PFBHA O-Oxime Yields (n=3)

Compound	MW (g/mol)	D (g/mL)	Y(± SD) (%)	R_TGC (min)	R_TLC$_I$ (min)	R_TLC$_{GE}$ (min)	CV Ia	CV Ir	RRF (SD)	TWC
Acetaldehyde	44.05	0.78	98.29(0.23)	5.77/ 5.89		14.76/ 14.47	9.0	22	44.2(0.7)	0.20
Acetone	68.03	0.79	100.00(0.15)	6.51	14.67	18.64	18	29	55(1)	1.7
Acetophenone	120.32	1.02	99.70(0.11)		39.79	32.56/ 27.79	22	12	96(2)	1.6
Acrolein	56.05	0.84	85.43(0.36)	7.03	15.31	19.73	11	29	41.9(0.9)	4.6[a]
Benzaldehyde	106.12	1.044	97.99(0.22)	23.20	30.79	29.54/ 26.82	24	28	100(2)	<0.04
n-Butanal	72.11	0.80	99.33(0.20)	8.97	23.02	26.20	12	9.0	44(1)	2.3
n-Crotonaldehyde	70.12	0.86	90.55(0.21)			24.06/ 22.82	29	70	41(1)	0.09
n-Decanal	156.27	0.83	99.54(0.23)	32.85	112.57	51.78/ 52.11	10		35(3)	2.5
Formaldehyde	30.03	0.82	99.64(0.10)	4.59	9.24	11.83	13	13	35.0(0.5)	5.5[a]
Glyoxal	58.04	1.14	84.45(0.43)	23.58	67.71	37.40/ 36.86	24	18	52.4(3)	1.1
n-Heptanal	114.19	0.818	99.27(0.10)	14.52	75.52	38.79/ 38.07	12	13	54.4(0.9)	14
Methyl glyoxal	72.06	1.045	88.51(0.22)	33.69	68.88[b]	41.93/ 41.32	3.0	23	50.2(4)	1.3
n-Valeraldehyde	86.13	0.81	99.37(0.31)	9.64/ 9.75		30.66	13	28	51.5(0.8)	0.90

[a], Maximal because of interfering peaks
[b], the monoderivative had a retention time of 14.67 min.

MW, Molecular weight; D, Density; Y, Yield corrected for purity; SD, Standard deviation; R_T, Retention time; GC, Gas chromatography/electron capture detection; LC_I, Liquid chromatography optimized isocratic method at 200 nm; LC_{GE}, Liquid chromatography optimized gradient elution method at 200 nm. More than one R_T signifies *E*- and *Z*- isomers with the peak of largest area given first; CV, coefficient of variation in % for 100 ng/mL at 200 nm in the optimized gradient elution LC method; Ia, intrarun; Ir, interrun; RRF, relative response factor at 200 nm on a molar basis relative to benzaldehyde as 100% in the optimized gradient elution LC method; and TWC, Los Angeles tap water concentration in ng carbonyl compound/mL tap water.

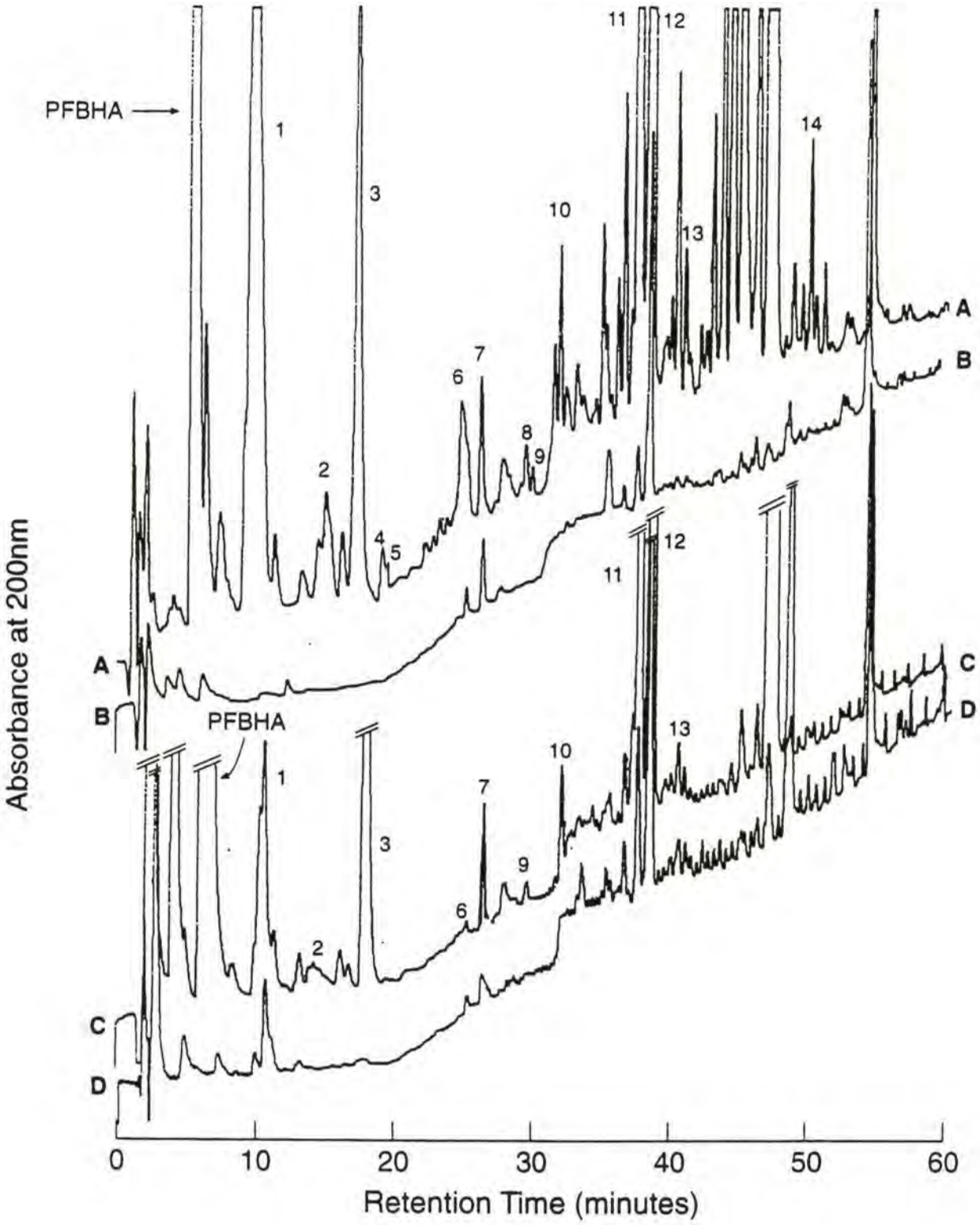
Table 2. Steric Dependence of Selected Carbonyl Compounds Relative to Synthetic Yield

Carbonyl Compound	Yield Corrected for Purity/Purity (n=3) (%)
Diisobutylketone	0.2(0.2)/5(5)
2-Heptanone	85(4)/99.0(0.5)
2,4-Hexanedione	48(3)/68(2)
Isobutyraldehyde	75.4(0.6)/99.0(0.1)
2-Methyl cyclohexanone	89.7(0.5)/98(1)

The quantities in brackets are standard deviations

LEGENDS FOR FIGURES

Figure 1. Gradient elution HPLC/UVD chromatograms at the same linear solvent gradient and temperature conditions at 200 nm (acetonitrile:water at 39 °C and 0.80 mL/min: 47.5% acetonitrile at 0 min; 53% at 16.9 min; 60% at 22.5 min; 65% at 28 min; 73% at 36.70 min; 75% at 38 min; 80% at 45 min; 95% at 60 min; and 100% at 70 min) for 500 mL of water (10 mL 0.1N sodium thiosulfate added) with and without PFBHA at pH 2.0, followed by addition of 5 mL 18N sulfuric acid, extraction with hexane (3 x 10 mL), evaporation of the solvent, and solid residue reconstitution in 0.60 mL of acetonitrile before priming a 10 μL injection loop with 50 μL of concentrate. A and B are tap water with and without PFBHA, respectively. C and D are distilled water with and without PFBHA, respectively. The identities of the PFBHA O-oxime peaks are provided for the original carbonyl compounds as follows: 1, formaldehyde; 2, acetaldehyde; 3, acetone; 4, acrolein; 5, *n*-crotonaldehyde; 6, *n*-butyraldehyde; 7, benzaldehyde; 8, *n*-valeraldehyde; 9, acetophenone; 10, glyoxal; 11, *n*-heptanal; 12, methyl glyoxal; and 13, *n*-decanal.


PFBHA
1
2
3
4
5
6
7
8
9
10
11
12
13
14
A
B
C
D
PFBHA
Absorbance at 200nm
0
10
20
30
40
50
60
Retention Time (minutes)

APPENDIX 6

A NEW PASSIVE SAMPLER FOR REGULATED WORKPLACE KETONES

SHIH-WEI TSAI[a] AND SHANE S. QUE HEE[b]

Department of Environmental Health Sciences and
UCLA Center for Occupational and Environmental Health
UCLA School of Public Health
10833 Le Conte Avenue
Los Angeles CA 90095-1772

[a]: Present Address: Department of Occupational Safety and Health
China Medical College
No. 91, Shiu-Shih Road, Taichung Taiwan
Republic of China

[b]: Correspondence author

NIOSH/CDCP Grant 5 RO1 OH03120 and the UCLA Center for Occupational and Environmental Health were responsible for financial support

ABSTRACT

A new solid sorbent passive air sampler for ketones has a silicone membrane atop a diffusion cylindrical path length of 1.1 cm and diameter 1.3 cm above a pellet of Tenax TA coated with 10 % (w/w) O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA). Vapors of known concentrations approximating their workplace permissible exposure limits of OSHA-regulated ketones at relative humidity (RH) of 3±1% were dynamically generated by a syringe pump connected to a dynamic air dilution system connected to an exposure chamber that allowed measurement of face velocities, temperatures, exposing vapor concentrations, and RHs. The O-oxime derivative was desorbed with hexane, and an aliquot injected for gas chromatographic analysis on a nonpolar capillary column by mass spectrometric or electron capture detection. The experimental passive sampler sampling rates in mL/min at 25 °C were 4.07±0.49, cyclohexanone; 6.30±0.59, diethyl ketone; 6.31±0.31, ethyl *n*-butyl ketone; 3.78±0.25, methyl *n*-amyl ketone; 3.43±0.19, methyl *n*-butyl ketone; 6.48±0.64, methyl ethyl ketone; 4.37±0.43, methyl isopropyl ketone; and 4.57±0.17, methyl *n*-propyl ketone. These preliminary data show that sterically unhindered ketones can be sampled by the passive sampler as well as aldehydes.

Keywords: passive sampler; ketone; personal sampling; adsorption; oxime; gas chromatography

INTRODUCTION

Ketones (R_1-(C=O)-R_2, where R_1 and R_2 are alkyl, aromatic, or alicyclic functional groups) are widely used industrial chemicals. They are used as solvents, chemical intermediates, cleaning fluids, dewaxers, and reaction enhancers, as well as in paints, hydraulic fluids, cleaning fluids, inks, pharmaceuticals, cosmetics, and dopes[1,2]. Ketones are also environmental products of photooxidation. For example, methyl ethyl ketone can be produced in outdoor air by the photooxidation of such air pollutants as butane and other hydrocarbons[3] . Methyl ethyl ketone has also been found in drinking water and surface waters[4]. Commercially important ketones include acetone, diacetone, methyl ethyl ketone, methyl *n*-propyl ketone, and methyl isobutyl ketone[2]. Ketones are mucous membrane irritants, but are not as potent as their closely related aldehyde analogs of the same number of carbon atoms. Ketones dehydrate the skin on contact. Overexposure can cause narcosis, headache, nausea, light-headedness, dizziness, and incoordination. Methyl *n*-butyl ketone is oxidized to the same neurotoxic metabolite (2,4-hexanedione) as is *n*-hexane, and peripheral and central neuropathy are caused in rats after TWA exposure to 1,300 ppm[2].

Methods to sample ketone vapors usually involve dynamic air sampling with solid sorbents[5]. NIOSH recommends several methods like charcoal tube sampling for acetone, cyclohexanone, diisobutyl ketone, 2-hexanone, methyl isobutyl ketone, and 2-pentanone[6]. However, CS_2 desorption of the more nonpolar ketones on charcoal tubes

is inefficient. A desorbing mixture of 1% methanol in CS_2 improves desorption of camphor, mesityl oxide, 5-methyl-3-heptanone, methyl-(*n*-amyl) ketone, and ethyl butyl ketone[7]. Methyl ethyl ketone is sampled on beaded carbon before desorption by CS_2[8]. 2-, 3- ,and 4- methyl cyclohexanone are sampled on Porapak Q, desorbed with acetone, and analyzed by GC[9]. Thermal desorption from graphitized carbon and carbon molecular sieves is used for ppb concentrations of ketone vapors[10].

The 2,4-dinitrophenylhydrazine (2,4-DNPH) solid sorbent method is recommended by the United States Environmental Protection Agency[3] to determine aldehydes and ketones in ambient air. The 2,4-DNPH method potentially allows relatively selective quantitation of different aldehydes and ketones through high performance liquid chromatography (HPLC)/ultraviolet (UV) detection of their hydrazones but not by GC since many hydrazones decompose at high temperatures. 2,4-DNPH does not react quantitatively with conjugated aliphatic aldehydes, can be light sensitive, and variable recoveries occur for liquid spiking[5].

The advantages of passive samplers[11] include lower cost and greater wearer acceptability as no bulky, expensive pumps are required. Some passive samplers have been developed for the lower molecular weight aldehydes and ketones based on liquid systems[12,13]. Passive sampling based on solid sorbents coated with 2,4-dinitrophenyl-hydrazine (DNPH) are available[14-17].

O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) has been used to analyze aldehydes in water because of its fast quantitative reaction to form O-oximes suitable for detection at the picogram (pg) level by gas chromatography/mass spectrometry (GC/MS) and gas chromatography/electron capture detection (GC/ECD) [18]. The PFBHA method also has been used to chemisorb aldehydes vapors by dynamic sampling[19,20] and passive sampling[21,22]. Both methods are not influenced by relative humidity (RH), temperature, intermittent sampling, shelf life for at least 3 months, and sample stability for at least 6 months. The aldehyde passive sampling method has now been extended to the major regulated positive interferences, the ketones.

EXPERIMENTAL METHODS

Materials

The ketones from Aldrich, Milwaukee , WI were: cyclohexanone (99.8%), diethyl ketone or 3-pentanone (99+%), *n*-butyl ethyl ketone or 3-heptanone (98%), methyl *n*-amyl ketone or 2-heptanone (98%), methyl *n*-butyl ketone or 2-hexanone (98%), methyl ethyl ketone or 2-butanone (99+%), methyl isopropyl ketone or 3-methyl 2-butanone (99%), and methyl *n*-propyl ketone or 2-pentanone (99.5%). Internal standard decafluorobiphenyl (99%) was also from Aldrich. Hexane (Optima), methanol (Optima), nitric acid, activated charcoal, molecular sieves, and indicating Drierite were from Fisher Scientific, Tustin, CA. O-(2,3,4,5,6-Pentafluorobenzyl)hydroxylamine hydrochloride or PFBHA was from Lancaster Laboratories, Lancaster, PA. Tenax TA

(80/100 mesh) was from Alltech Associates, Deerfield, IL. Helium, nitrogen, and 5% methane/argon, all chromatographic grade, were from Alphagaz, Los Angeles, CA.

Equipment

Pyrex tubing, Pyrex glass wool, 4-mL Kimble vials with PTFE-lined screw caps, 10-μL Hamilton syringes, gas-tight Hamilton syringes, Soxlett-extraction apparatus, a Parr 2811 bench manual pellet press, 3M Model 3500 OVM passive sampler, Bel-Art clear polycarbonate vacuum desiccator that served as exposure chamber, calibrated temperature/relative humidity (RH) meter/recorder, hot/cold hair dryer, a Thermolyne Series 5000 carbon dioxide incubator for elevated temperature experiments, and Harvard syringe pumps (model 11) were from Fisher Scientific. Personal sampling pumps, rotameters, and Tedlar gas bags were from SKC West, Fullerton CA. A Whatman Zero Air generator was from Balston, Haverhill, MA. A M-5 Mini-Buck calibrator for flow rate measurement was from Buck Scientific, East Norwalk CT. A Goldstar Multiwave shelf microwave oven for O-oxime syntheses was from Circuit City, Westwood CA. A small box desk air fan connected to a variac to generate constant face velocities in the exposure chamber was from Tekna Design, Rockford MI,

GC/MS was done with a Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, Calif.) equipped with a 30-m × 0.32-mm ID 1 μm film DB-1701 chemically bonded fused-silica capillary column. The temperature for the injector and link was

250°C. The column temperature program was: solvent delay 5 min at 105°C, 105°C for 0.5 minutes, 105°C to 220°C at 10°C/minute, and holding then for 10 minutes. The Hewlett-Packard 5988A quadrupole mass spectrometer had an electron multiplier detector, and the 70 eV ion source temperature was 260°C. Selective ion monitoring (SIM) used m/z 181 and total ion monitoring (TIC) m/z utilized 50-500. The areas of both *E*- and *Z*- isomers were utilized for quantitations for asymmetric ketones.

The same column and temperature conditions were used for Hewlett-Packard 5890 capillary GC/^{63}Ni-electron capture detection (ECD) with 5% methane/argon flow of 3.0±0.4 mL/min. The detector temperature was 250°C. The signal was visualized with a Hewlett-Packard 3396 integrator.

Methods

A 13 mm diameter and 0.3 cm thick pellet of PFBHA coated Tenax GC (10%, w/w) was made by the hand press, after the coating and drying procedure described elsewhere[21]. The sampler utilized the silicone membrane, badge body, and badge clip unchanged from the 3M Model 3500 OVM sampler[21]. The pellet was placed into a Teflon-lined screw cap of dimensions 18-mm o.d., 14-mm i.d., internal depth 14 mm, and outer height 16 mm. In its field form, the screw cap was secured centrally to the bottom of the empty 3M badge body by a small piece of duct tape. The 3-prong stay of the 3M sampler was cut to allow the pellet to be held securely by one of its prongs.

The 3M silicone membrane and then a 10-μm Teflon filter of the same diameter was placed over it and both fixed tightly over the cap with an aluminum seal via a crimper. The whole sampler was wrapped in aluminum foil (shiny side out) until sampling, which was initiated by removing the foil. The diffusion path length was 1.1 cm (Figure 1). The vapor generator, air dilution system, and exposure chamber (Figure 2) are described in detail elsewhere[21,22]. The air generator was connected to the vapor and water generation sites to provide constant defined flow rates. Vapor generators were syringe pumps set at known plunger velocities to produce the desired concentration of organic vapor for dilution, or water vapor for RH. Heating tape wrapped around the outside of the stainless steel tubing at the needle exit from the syringe pumps ensured total volatilization of organic vapor or water. The two streams were then routed through a stainless steel T-joint adapter, and the outlet connected by Teflon tubing to a Greenburg-Smith impinger which acted as a mixing chamber. Teflon tubing then conveyed the diluted organic vapor into the exposure chamber through a hole bored on the side near the chamber bottom to just underneath the fan blades, the fan resting at the bottom of the chamber and under the ceramic metal plate. Six samplers were set horizontally on the plate each with a nearby closable hole in the chamber wall for probe insertion for measurement of RH, temperature, organic vapor concentration, simultaneous dynamic sampling[34] using Tenax TA coated with 20% Tenax TA[19], and face velocity.

Synthesis of PFBHA O-Oximes

The PFBHA O-oximes are not commercially available. They were synthesized by methods detailed elsewhere[24]. The major difference in the synthetic method for the ketones relative to the aldehydes[19,22,23] was to allow the reacted solution to cool to room temperature gradually rather than by ice bath cooling. This procedure increased ketone O-oxime yields[24].

Ketone Diffusion Coefficients and Sampling Constants

The dependence of the diffusion constant on molecular weight and temperature is expressed through equation (1)[25]:

$$D_{AB} = \frac{0.00143 \times T^{1.75}}{P M_{AB}^{1/2} [(\sum_v)_A^{1/3} + (\sum_v)_B^{1/3}]^2} \qquad (1)$$

where D_{AB} is the binary diffusion coefficient of analyte in air in cm^2/s at T

T is temperature, K

M_A and M_B are molecular weight, g/mol

$M_{AB} = 2[(1/M_A)+(1/M_B)]^{-1}$

P is the external pressure, bar

Σ_v is the summation of atomic diffusion volumes, unitless

i is all the contributing species

A is air

B is the analyte

The molecular diffusion volume of air is 19.7[25].

For example, the Σ_v , for cyclohexanone, $H_{10}C_6O$, is calculated as:

$$(\Sigma_v)_{H_{10}C_6O} = 10\times2.31^{H} + 6\times15.9^{C} + 1\times6.11^{O} = 124.61 \quad (2)$$

where H = 2.31 is the atomic diffusion volume increment for hydrogen

C = 15.9 is the atomic diffusion volume increment for carbon

O = 6.11 is the atomic diffusion volume increment for oxygen

Then, the diffusion coefficient of cyclohexanone at 25 °C and 1 atm (1.01 bar) is:

$$D_{Air\text{-}Cyclohexanone} = 0.00143\times298^{1.75}/\{1.01\times[2\times(1/28.8+1/98.14)^{-1}]^{1/2}\times[19.7^{1/3} + 124.61^{1/3}]^{2}\} = 0.0766\ cm^2/s \quad (3)$$

The theoretical sampling constant k is in Fick's first law of diffusion as shown in equation (4) in its form for a cylindrical open tube[26]:

$$dm/dt = (D_{AB}A/L)(c_{air}-c_{surf}) = k(c_{air}-c_{surf}) \quad (4)$$

where dm/dt is the steady state mass sampling rate or mass transfer rate, weight/time

A is the effective cross-sectional area of the sampling element, cm^2

L is the effective path length where diffusion control prevails to the sampling element from the exposing atmosphere, cm

c_{air} is the air concentration of the analyte, weight/cm^3

c_{surf} is the air concentration of analyte just above the sampling surface in the same units as c_{air}

k is the sampling constant of the analyte, (D_{AB} A/L), cm^3/time

For the sampler, A/L is $(\frac{1.3}{2})^2 \times \pi \times \frac{1}{1.1} = 1.2$ cm.

Therefore the sampling constant k for cyclohexanone is:

$k = D_{AB}A/L = 1.2\ D_{AB} = 0.092\ cm^3/s$

Reaction Efficiency/O-Oxime Recovery for Wet Spiking of Ketones

Liquid ketone equivalent to two times the PEL-8 hour mass as determined from the theoretical sampling constant was spiked with 50 μL of methanol solution. The spiked pellet was held overnight in a desiccator containing Drierite to allow the methanol to dry before desorption with 2.0 mL hexane at room temperature over 2 hours with 30 sec of ultrasonication at every half-hour before analysis by GC/MS or GC/ECD using synthesized aldehyde O-oximes of known purity.

Face Velocity, Relative Humidity and Temperature

For all experiments, the face velocities were above 20 fpm (0.10 m/s), the critical face velocity[21]. The range of face velocities in a typical workplace is from 20-30 fpm[11]. The RH was 3±1%, and the temperature was 22±1°C. The low RH and the single temperature were selected since previous work with the aldehydes acrolein and n-valeraldehyde had shown no dependence of sampling constants on RH and temperature[21]. All data were corrected to 25°C and 1 atmosphere pressure using the Ideal Gas Law and observed temperatures and atmospheric pressures.

Vapor Exposures

The ppm-hour levels of exposure for dynamic and passive sampling were equivalent to 0, 0.5, 1.0 and 2.0 times the PEL for 8 hours. Previous work showed that the absolute recovery for valeraldehyde vapor in the dynamic method varied with flow rate, 10 mL/min being better (efficiency of about 100%) than 50 mL/min (efficiency 71-85%)[19-21]. Therefore the flow rate for sampling pumps in the dynamic sampling method was set at 10 mL/min because the contact and reaction times were expected to be important for the dynamic sampler for ketones. The dynamic sampling technique was utilized as the reference method after its sampling efficiency was determined for each ketone using the static gas bag method. This entailed injecting a known volume of liquid ketone into the Tedlar gas bag containing a known volume of pure or prehumidified air, desorbing with hexane, and then GC/ECD or GC/MS analysis, as described at length in reference

19[34]

Previous passive sampling work with the aldehydes acrolein and n-valeraldehyde had shown that exposures to TLV-TWA concentrations for 8 hr were equivalent to ppm-hr exposures when exposed in 1-hr exposures using 3 different intermittent exposure regimens over 8 hr (8 times the target concentration for 1 hr, the same followed by 7 hr of exposure to pure air, or three 1-hr exposures separated by two 1-hr exposures to pure air). Thus the concentrations used for each ketone were at 0, 4, 8 and 16 times the appropriate PEL for 1-hr exposures to obtain the desired ppm-hr exposure doses.

The O-oxime in the pellet was desorbed with 2-mL hexane, and analyzed by GC/MS or GC/ECD on injection of 2 μL. The μmoles desorbed corrected for desorption efficiency was plotted against (μmole/mL)×min to provide k as the slope of the linear regression line from the 4 concentrations generated for each ketone.

Statistics

All internal comparisons were subjected to analysis of variance types I and II to detect significant differences at $p \leq 0.05$ and significant interactions[27]. Linear regression analyses included calculation of standard deviations of the slopes and intercepts.

RESULTS

Table I shows the yields for O-oxime syntheses corrected for GC/MS purities. The latter correct for the presence of pentafluorobenzaldehyde, pentafluorobenzyl alcohol, excess PFBHA in the O-oximes, any other aldehydes, and other peaks not attributable to the reagents and solvents[23]. All yields are greater than 97.8%, based on the 1:1 stoichiometry. The average purity was 98.40±0.36%. Methyl isopropyl ketone yield exceeded 75% though that for diisopropyl ketone did not[24].

Table I also shows the results of reaction efficiency/O-oxime recovery for wet and vapor spiking of ketones relative to pure PFBHA-ketone standards. The lowest wet spiking efficiency was shown by cyclohexanone at 87% which also had the highest coefficient of variation of 8.0%. The results are similar to those previously observed for aldehydes where no significant statistical difference at $p \leq 0.05$ was observed between O-oxime standard curve analysis and wet aldehyde spiking[21]. The detection limits for each O-oxime were between 110-200 pg at 2 times the background.

The results of dynamic sampling for known gas bag vapor concentrations at flow rate 10 mL/min are also shown in Table I. Vapor sampling efficiencies all exceeded 78.9%. The vapor spiking recoveries lower than for wet spiking at $p \leq 0.05$ were for *n*-butyl ketone, methyl isopropyl ketone, and methyl *n*-propyl ketone, with the lowest recovery being for methyl isopropyl ketone. Acetone with its PEL of 1000 ppm

exceeded the capacity of the sampler, though it produced O-oxime derivatives efficiently by wet chemistry[24].

Table II shows the passive sampling constants k in equation (4) from the results of linear regressions of the ketone vapor moles collected for four concentrations versus the ketone concentration × time curves. The coefficient of variation (CV) of the slope was $< 10\%$ for all ketones except cyclohexanone where it was 12%. All intercepts were not significantly different from zero at $p \leq 0.05$ except for diethyl- and methyl ethyl-ketones. Thus only the latter two ketones do not obey a Henry type law over the ppm-hr range investigated.

Table II also provides the theoretical D and k values at 25°C and 1 atm from equations (2) and (4), respectively, in addition to the experimental D, and to the sampling efficiencies relative to theoretical (equations (2) and (4)). While diethyl-, ethyl *n*-butyl-, and methyl ethyl- ketones did not give D values different from theoretical, those for cyclohexanone and methyl *n*-amyl-, methyl isopropyl-, and methyl *n*-propyl- ketones gave experimental values between 73-77% of those predicted. The experimental value for methyl *n*-butyl ketone was about 63% of that predicted. Except for methyl *n*-butyl ketone, all the experimental sampling constants for ketones tested were within ±25% of the theoretical sampling constants. Why ethyl *n*-butyl ketone has a higher relative efficiency is unknown. Therefore, D values must be verified experimentally.

DISCUSSION

The average experimental D relative to theoretical D from equation (4)(Table II) for all the eight ketones studied was 86±21%, compared with 102±28% for aldehydes[22]. This probably reflects steric effects in ketones because the aldehyde hydrogen is small relative to the second ketone alkyl group.

For example, diethyl ketone, methyl isopropyl ketone, and methyl *n*-propyl ketone were all $C_5H_{10}O$ with different carbon chain structures. The propyl group caused lower relative efficiencies of between 74-77% (Table II) whereas diethyl ketone did not. The same behavior was observed in the dynamic sampling results. The methyl *n*-butyl ketone also had low relative efficiencies in dynamic and passive sampling (Table II). Ethyl *n*-butyl ketone and methyl *n*-amyl ketone were both $C_7H_{14}O$ with the *n*-amyl group causing a lower relative efficiency of 73-75% for passive sampling, that was not observed for dynamic sampling, the same being so for cyclohexanone.

The ketones selected here were relatively sterically unhindered since it is known that when chain branching occurs at both carbons β- to the carbonyl carbon, even the wet chemistry synthesis is drastically inhibited for diisobutylketone and 2,4-hexanedione [24] Nevertheless, the gas phase/solid phase recoveries are sufficiently high and precise to show that the PFBHA passive and dynamic samplers developed for aldehydes will also function well for these sterically unhindered ketones, and also confirms that the

ketones may potentially act as positive interferences to aldehydes for this PFBHA air sampling method. Further work is required to assess the sampler for more sterically hindered ketones, and to confirm whether RH, temperature, and intermittent sampling effects are negligible as found for the aldehydes.

The major alternative passive air sampler based on 2,4-dinitrophenylhydrazine (DNPH) has been validated for formaldehyde, acetaldehyde and glutaraldehyde[14,15,28,29]. Little attention has been paid to the factors that affect 2,4-dinitrophenylhydrazone HPLC analysis until recently[30]. Ozone is also known to interfere with the DNPH coated silica gel air sampler reaction with formaldehyde[31] in ambient air sampling. This remains to be tested for the present passive sampler. More sensitivity (110-200 pg) and selectivity (selective ion monitoring by GC/MS with m/z 181) are possible for GC/MS and GC/ECD using PFBHA O-oxime derivatives than with HPLC/UVD or liquid chromatography/mass spectrometry (LC/MS) for the DNPH method[18,32,33], thus allowing unknowns to be better assigned and quantified. The five fluorine atoms in the PFBHA O-oximes allow sensitive screening GC/ECD[18].

The advantages of a predominantly GC method over a HPLC one are many: there are many laboratories equipped with flame ionization detectors (FID) and ECD GCs, whereas HPLC entails much waste disposal and LC/MS is still not as available as GC/MS. GC/MS allows for specific identification of carbonyl (e.g. aldehydes and

ketones) derivatives within 30 min, whereas HPLC separation of O-oximes takes up to 120 min[24].

The present study has shown that a passive sampler and a dynamic sampler at flow rate 10 mL/min designed for aldehydes with PELs up to 50 ppm also is adequate for sterically unhindered ketones with PELs up to 200 ppm. The results imply that acetone vapor TWA sampling is possible up to 200 ppm, though at 1000 ppm the capacity is exceeded.

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TABLE I. Efficiencies from Wet Syntheses, Wet Spikings, and Dynamic Sampling in Terms of O-Oxime Recovered.

Ketone (OSHA PEL in ppm, Yield±SD)	Wet Spiking(%)	Dynamic Sampling(%)
Cyclohexanone (50, 98.4±1.6)	87.2 ± 7.0	95.5 ± 0.11
Diethyl Ketone (200[a], 98.91±0.17)	91.1 ± 3.9	95.6 ± 8.5
Ethyl *n*-Butyl Ketone (50, 98.3±2.8)	98.0 ± 3.2	97.9 ± 3.2
Methyl *n*-Amyl Ketone (100, 98.7±1.1)	93.3 ± 2.7	93.5 ± 4.5
Methyl *n*-Butyl Ketone (100, 98.67±0.14)	107.00 ± 0.14	83.8 ± 4.1
Methyl Ethyl Ketone (200, 97.8±1.4)	90.0 ± 4.0	100.1 ± 6.7
Methyl Isopropyl Ketone (200, 98.33±0.36)	92.8 ± 5.6	78.9 ± 4.8
Methyl *n*-Propyl Ketone (200, 98.1±1.3)	92.5 ± 2.6	83.8 ± 4.3

Average (NA, 98.4±0.36)	93.9±6.1	91.1±7.8

[a], No OSHA PEL: 1998 ACGIH TLV-TWA; NA, Not Applicable.

The ± quantities are standard deviations for n=3.

Yields are relative to 1:1 stoichiometry, and correct for GC/MS purities

TABLE II. Theoretical and Experimental Passive Sampling Constants

Ketone	Theoretical Diffusion Coefficient (cm^2/s)	Theoretical Sampling Constant (cm^3/min)	Experimental Diffusion Coefficient (cm^2/s)	Experimental Sampling Constant $cm^3/min(r^2)$	Relative Efficiency [A] (%)
Cyclohexanone	0.077	5.59±0.43	0.0562±0.0067	4.07±0.49(0.972)	73.1±9.1
Diethyl Ketone	0.082	5.97±0.46	0.0869±0.0081	6.30±0.59(0.983)	106±11
Ethyl *n*-Butyl Ketone	0.069	5.03±0.39	0.087±0.0043	6.31±0.31(0.995)	126±10
Methyl *n*-Amyl Ketone	0.069	5.03±0.39	0.0521±0.0034	3.78±0.25(0.992)	75.4±6.7
Methyl *n*-Butyl Ketone	0.075	5.46±0.42	0.0472±0.0026	3.43±0.19(0.994)	63.1±5.2
Methyl Ethyl Ketone	0.092	6.68±0.51	0.0892±0.0088	6.48±0.64(0.981)	97±10
Methyl Isopropyl Ketone	0.082	5.97±0.46	0.0601±0.0059	4.37±0.43(0.981)	73.5±7.9
Methyl *n*-Propyl Ketone	0.082	5.97±0.46	0.0628±0.0023	4.57±0.17(0.997)	76.8±5.7

[A], Relative Efficiency (%) = [(Experimental Sampling Constant/Theoretical Sampling Constant from equation (4)]×100%

Figure Captions

Figure 1. Cross Section of the PFBHA Passive Sampler

Figure 2. Ketone Vapor Generation and Exposure Chamber System

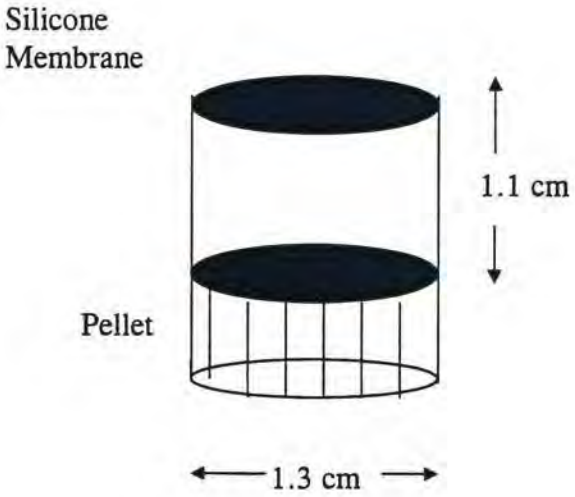


Diagram of Sampler

Fig. 1.

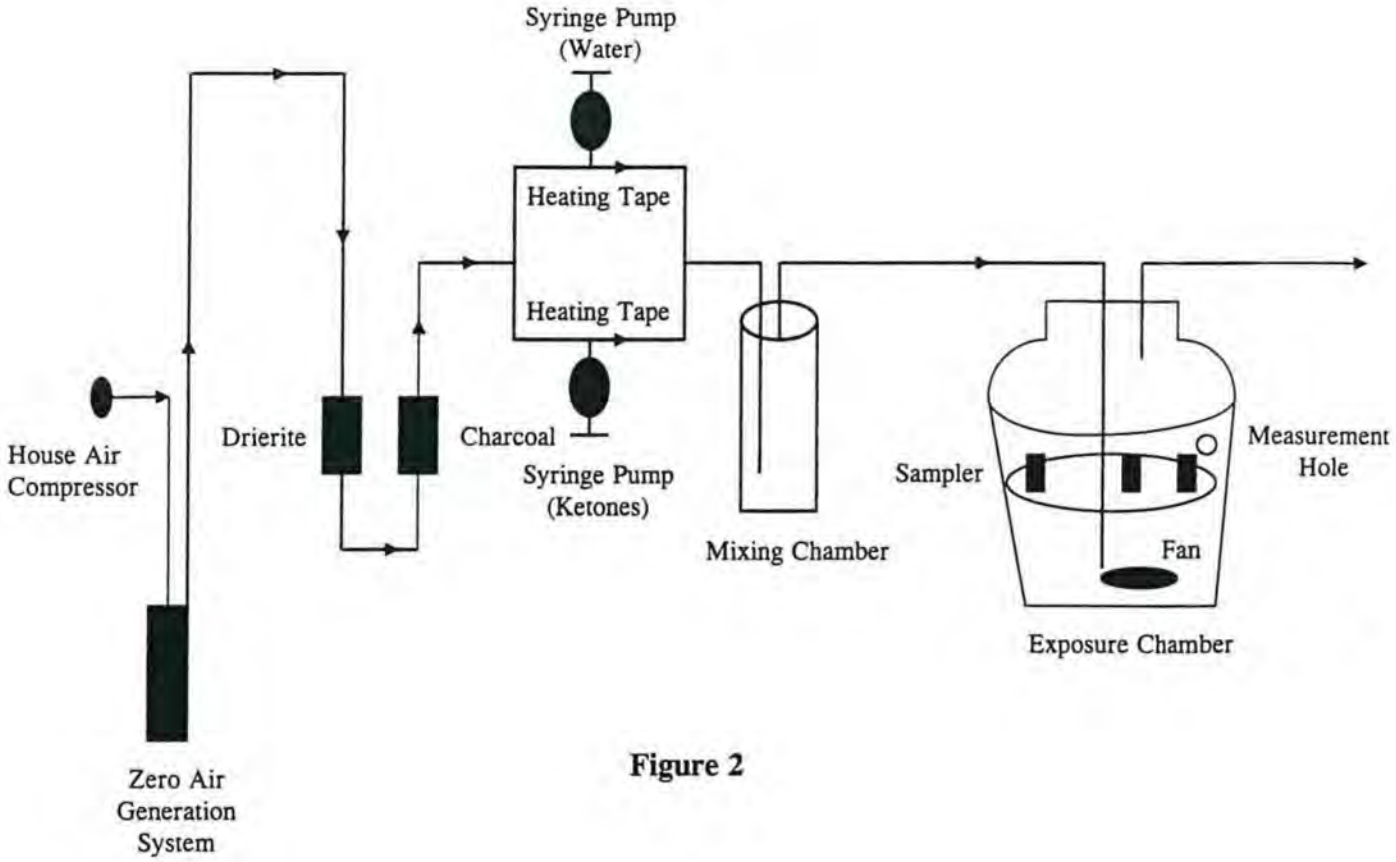

Syringe Pump
(Water)
Heating Tape
Heating Tape
House Air
Compressor
Drierite
Charcoal
Syringe Pump
(Ketones)
Mixing Chamber
Sampler
Measurement
Hole
Fan
Exposure Chamber
Zero Air
Generation
System

Figure 2

APPENDIX 7

VAPOR CHEMISORPTION CLASSICAL ISOTHERMS OF

A SOLID SORBENT PASSIVE SAMPLER

FOR ALDEHYDES

SHIH-WEI TSAI AND SHANE S. QUE HEE*

Department of Environmental Health Sciences and
UCLA Center for Occupational and Environmental Health
UCLA School of Public Health
10833 Le Conte Avenue
Los Angeles CA 90095-1772
Phone: (310)206-7388; Fax: (310)794-2106; E Mail: squehee@ucla.edu

NIOSH/CDCP Grant 5 RO1 OH03120 and the UCLA Center for Occupational and Environmental Health were responsible for financial support

ABSTRACT

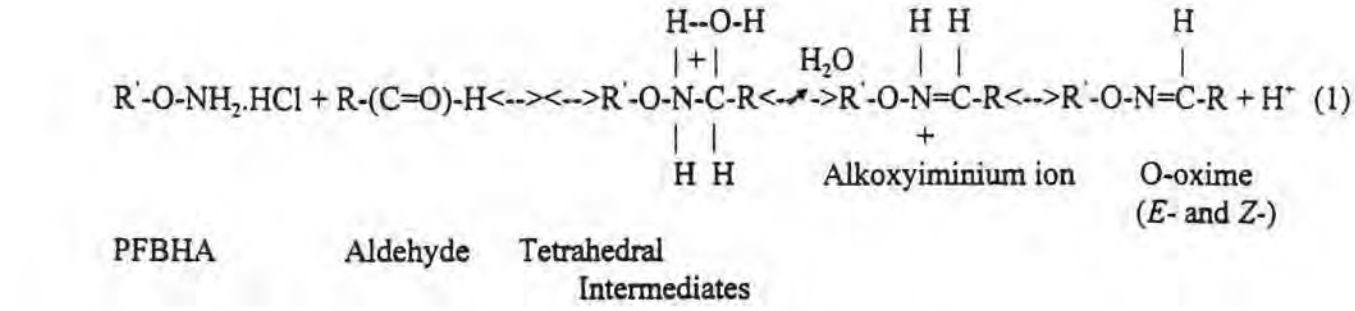
The vapor-phase chemisorption isotherms of 1-pentanal (*n*-valeraldehyde) and 2-propenal (acrolein) above the critical face velocity (15-20 fpm; 7.6-10 cm/s) for a passive air sampler based on O-(2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (PFBHA) coated Tenax solid sorbent were investigated at 25 °C. The sampler had a silicone membrane atop the sampling cylinder of diffusion path length 1.1 cm and diameter 1.3 cm above the chemisorption pellet of 10% (w/w) PFBHA on Tenax TA (80/100 mesh). Vapors of known concentrations and at specific relative humidities (RH) were generated by the syringe pumps of a dynamic generation and dilution system. An exposure chamber allowed measurement of face velocities, temperatures, exposing vapor concentrations, and RH. The O-oxime derivatives were desorbed quantitatively with hexane, and an aliquot injected for gas chromatographic analysis on a nonpolar capillary column by mass spectrometric or electron capture detection. The pellet capacity was about 30 μmoles. RH (3-79%) and temperatures (4-48 °C) had no effect on aldehyde chemisorption. Analysis of the isotherm data showed that 1-pentanal adsorption could be best fitted by a Langmuir-BET I model, and that for acrolein was described best by a Dubinin-Radushkevich model. The connections between the models were discussed. Tortuosity and available air space and the partition coefficient within the microporous pellet were important parameters to explain why the classical models did not predict experimental saturation capacities. The activation energy analysis was suggestive of physical adsorption and/or hydrogen bonding being the rate determining step rather than addition across the double bond.

Aldehydes (R-(C=O)-H where R is alkyl, aromatic, or alicyclic) are not only important industrial chemicals, but they are also ubiquitous volatile products of combustion and oxidation[1-3]. Methods for the analysis of aldehyde vapors usually use solid sorbent dynamic air sampling[1-7]. The advantages of passive samplers are well known[8,9], and the latter have been developed for the lower molecular weight aldehydes and ketones based on solid sorbents coated with 2,4-dinitrophenylhydrazine [10-13]. No adsorption isotherm studies exist for passive chemisorption applications of air sampling.

Sensitive gas chromatography (GC)/mass spectrometry(MS) and GC/electron capture detection (ECD) methods can be used to determine the O-oxime derivatives of O-(2,3,4,5,6-pentafluoro)-benzylhydroxylamine hydrochloride (PFBHA) in dynamic[7] and passive [14,15] air sampling. In both dynamic sampling with 200 mg of 20% PFBHA on Tenax GC[7] and passive sampling with a 150 mg 13.0-mm diameter pellet of 10% PFBHA on Tenax TA (Figure 1)[14,15], aldehyde vapor sampling is not affected by temperature and RH at a given flow rate for dynamic sampling, and above the critical face velocity for passive sampling. There is a small dependence on flow rate for dynamic sampling[7]. There are only small effects of RH on Tenax TA and Tenax GC retention volumes of organic adsorbates[16]. Vapors of liquid aldehydes and ketones are also retained on uncoated Tenax GC and TA[17,18]. Tenax sorbents have very high affinity for aromatics and halogenated compounds[16-18]. PFBHA O-oximes also show high affinity.

Since Tenax TA has a small retention volume for water vapor and the O-oxime is a solid and hydrophobic, the reaction in the presence of excess PFBHA is pulled over to the retained product

so that the moles of O-oxime equal the reacted moles of exposing carbonyl vapor. The actual mechanism of the gas phase/solid phase chemisorption reaction is unknown. Assuming classical carbonyl nucleophilic addition chemistry[19], the trigonal carbonyl group (bond length 0.120-0.122 nm and bond energy of about 670 kJ/mol[20]) reacts with one molecule of PFBHA to form several tetrahedral intermediates, and eventually the corresponding O-oxime[19] as in equation (1):



where R' is the (2,3,4,5,6-pentafluorobenzyl)- functional group.

The specific tetrahedral intermediate in equation (1) is one of many known to be formed. Its formal covalent σ C-O bond has an energy of about 340 kJ/mol and a length of 143 pm. The hydrogen bonds have energies between 13-32 kJ/mol[20]. The RCH[(O)H---N] bond is about 216 pm in length and 14.3 kJ/mol in energy in the cyclic form. Similarly, the RCH[(O)H---O] bond is between 193 pm and 29.1 kJ/mol, and between 190-205 pm and 21.7-31.6 kJ/mol for the open *E*- and *Z*- forms respectively. The intramolecular H-bond lengthens from 207 to 240 pm on external protonation and decreases variably in strength.

The actual addition process is usually visualized in two ways[19]: the carbonyl group and the electron pair of a strong nucleophile first form a tetrahedral oxyanion, which is further protonated

twice at the oxygen, and then dehydrated to produce O-oxime product. The neutral intermediate is probably the most stable one. The other mechanism is based on acid catalysis to protonate the carbonyl group first to give an oxonium ion, whose carbon accepts the electron pair of a weak nucleophile to produce the first tetrahedral intermediate, with a hydrogen on the bonded nucleophile being then removed by a base to regenerate the acid and produce the most stable tetrahedral intermediate of the first mechanism. The common intermediate after both addition processes is an alkoxyiminium[19] ion formed from the protonated aminoalcohol tetrahedral intermediate in equation (1).

PFBHA in its hydrochloride salt form might act as a weak, intermediate, or strong nucleophile. A 4 mM PFBHA hydrochloride salt solution has an initial pH of 2.89[21]. The pK_a is 3.0-3.2[21]. Thus, PFBHA free amine is a weak base, and that its protonated hydrated conjugated acid is a relatively strong Lewis acid since the pH of 4 mM HCl is 2.40. The aqueous reaction has its optimum rate in the 1.0-3.0 pH range[21] where protonation exceeds 50%, compared with the pH range 4-5 for alkyl iminium ions[19]. Which mechanisms occur in the gas phase/solid phase aldehyde-PFBHA reaction are unclear, and will be investigated in the present study.

The PFBHA air sampling methods[7,14,15] are practical since the O-oxime product is less volatile than the original aldehyde, is retained on the surface, and on desorption can be sensitively analyzed. A large excess of PFBHA constitutes a large reactive capacity, and the O-oxime product mass is controlled by the carbonyl compound as the limiting reagent. The O-oxime is then desorbed for analysis rather than the original exposing compound as in physical adsorption.

The reaction stoichiometry allows linking of the determined mass of O-oxime to the original chemisorbed mass of aldehyde, after correction for desorption and sampling efficiencies.

Only the fraction of aldehyde molecules available to the PFBHA on the sorbent surface reacts in passive sampling. If the fraction is constant as occurs above the critical face velocity at constant exposure conditions, a sampling constant k can be calculated that links the O-oxime mass produced by chemisorption to the exposure concentration through Fick's first law of diffusion (equation (2)) in its form for a cylindrical open tube[14,15]:

$$dm/dt = (DA/L)(C_A - C_{surf}) = k\,(C_A - C_{surf}) \qquad (2)$$

where dm/dt is the steady state mass sampling rate or mass transfer rate, weight/time

D is the diffusion coefficient of the analyte in air, cm^2/time

A is the effective cross-sectional area of the sampling element, cm^2

L is the effective diffusion path length of the sampler, cm

C_A is the air concentration of the analyte, weight/cm^3

C_{surf} is the air concentration of analyte at the surface in the same units as C_A

k is the sampling constant of the analyte, equal to (DA/L), cm^3/time

If the time of exposure t is known, C_A is found by dividing the equivalent aldehyde mass by the volume of air sampled (kt in cm^3) assuming C_{surf} is zero. The volume is corrected to 25 °C and 760 mm Hg via the Ideal Gas Law.

The dependence of D on molecular weight and temperature is expressed through equation (3)[23]:

$$D = 0.001\ T^{1.75}(1/M_{Air} + 1/M_A)^{0.5}/\{p_e[(\Sigma_{Air}\, v_i)^{1/3} + (\Sigma_A\, v_i)^{1/3}]^2\} \qquad (3)$$

where D is the diffusion coefficient of analyte in air in cm^2/s

T is temperature, K

M_{Air} and M_A are molecular weights for air (Air) and the analyte (A) respectively, g/mol

p_e is the external presure, bar

v_i are normalized molecular diffusion volumes, unitless

i is all the contributing species

The present study is concerned with elucidating the mechanism of the chemisorption process of equation (1) through study of the aldehyde vapor/solid sorbent isotherm behavior.

EXPERIMENTAL

The two aldehydes investigated were acrolein (molecular weight 56.06, vapor pressure at 25°C is 274 mm Hg, boiling point at 76 cm Hg 52.5 °C, and liquid density at 20 °C is 0.843 g/mL) and *n*-valeraldehyde (molecular weight 86.13, vapor pressure is 50 mm Hg at 25 °C, boiling point at 76 cm Hg 102 °C, and liquid density at 11 °C is 0.818 g/mL), over the extremes of the 1996 Threshold Limit Values of 0.1 ppm and of 50 ppm over 8 hr, respectively. Though not commercially available, the PFBHA O-oximes have been synthesized[7].

Materials

The aldehydes were acrolein (97%) and valeraldehyde (99%) from Aldrich, Milwaukee, WI, as was the GC internal standard, decafluorobiphenyl. Hexane (Optima), methanol (Optima), nitric acid, activated charcoal, molecular sieves, and Drierite were from Fisher Scientific, Tustin, Los Angeles. PFBHA was from Lancaster Laboratories Inc., Lancaster, Penn. Tenax TA (80/100 mesh) was from Alltech Associates, Deerfield, Ill. It is a microporous 2,6-diphenyl-*p*-phenylene oxide polymer of nominal specific surface area of 35 m^2/g[18]. Helium, nitrogen, and 95% methane/argon were ultrapure grade from Alphagaz, Los Angeles, CA.

Equipment

The following were from Fisher Scientific: Pyrex tubing 7-mm OD and 5-mm ID broken into lengths of 7 cm with their ends fire-polished in a propane-air flame of a piezo electronic microtorch (Blazer Products, New York); Pyrex glass wool cleaned by methanol and hexane Soxhlett-extractions; 4-mL Kimble vials with PTFE-lined screw caps (Fisher 03-340-60A); Pyrex ground glass volumetric flasks, beakers, V-vials, round-bottom flasks, spatulas, test tubes, and pipets; propipets; 10-uL Hamilton syringes; gas-tight Hamilton syringes; Soxlett-extraction apparatus; bench centrifuges; Pasteur pipets; charcoal-lined respirator; vacuum desiccators; Buchi rotary evaporator; hot/cold air hair dryer; calibrated humidity/temperature meter/recorder; a Parr 2811 bench manual pellet press (Fisher Scientific 04-379); 3M Model 3500 OVM passive sampler; Bel-Art clear polycarbonate vacuum desiccator (Fisher Scientific 08-642-7) with a ceramic metal plate (Fisher 08-642-10); Harvard syringe pump (model 11) , screw-caps 14 mm ID and depth 16 mm, a carbon dioxide incubator (Thermolyne Series 5000), heating tapes,

Greenburg-Smith impingers; variacs, TFPE Teflon and Tygon tubing 6.4 mm ID, and a Mettler AE260 analytical balance. Personal sampling pumps (model P30A) were from DuPont. Tedlar gas bags from 10- to 100-L were from SKC Inc, Eighty Four, Penn. Stainless steel tubing of 6.4 mm OD, stainless steel swagelok and ferrule T-adapters were from Alltech Inc. A Vibro-Graver vibrator was from Burgess Vibrocrafters, North Adams, Mass. A M-5 Mini-Buck Calibrator for air flow rate calibrations was from Buck Scientific, East Norwalk, Conn. Solid sorbents were Soxhlett-extracted overnight with methanol, and then hexane before being dried for two hours at 80°C in an incubator, with final drying to constant weight in a vacuum desiccator. Pure air was supplied by a Whatman Zero Air generator from Balston Inc. A 11.6 eV photoionization organic vapor analyser was from H-Nu, Inc (model PI-101). Rotameters were from SKC Inc. A calibrated model 8500 II hot-wire anemometer was from Alnor Instrument Co. (Skokie, IL.). A small box desk fan for face velocity studies was part number 14244-01 from Tekna Design (Rockford, MI). A Goldstar Multiwave shelf microwave oven (Circuit City, Los Angeles, Calif.) facilitated PFBHA O-oxime syntheses.

GC/MS was done with a Hewlett-Packard 5890 gas chromatograph (Hewlett-Packard, Palo Alto, Calif.) equipped with a 30-m x 0.32-mm ID 1 μm film DB-1701 chemically bonded fused-silica capillary column (J&W Scientific, Folsom, Calif.) linked with the 70 eV electron impact ion source of a Hewlett-Packard 5988A mass spectrometer having a quadrupole mass filter and an electron multiplier detector. Injections of 2 μL hexane solution were at a helium carrier flow of 3.0±0.3 mL/min in the splitless mode with a purge delay of 1 min. The temperature for the injector and link was 250 °C. The column temperature program for valeraldehyde was: solvent

delay, 5 min at 105 °C; 105 °C for 0.5 minutes, 105 °C to 180 °C at 10 °C/minute, and holding then for 4 minutes. The column temperature program for acrolein was: solvent delay, 5 min at 105 °C; 105 °C for 0.5 minutes, 105 °C to 230 °C at 10 °C/minute, and holding then for 9 minutes. The ion source temperature was 260 °C. Selective ion monitoring (SIM) utilized m/z 181 and total ion monitoring m/z 50 through 500. The areas of both *E*- and *Z*- isomers were utilized for quantitations. Linear ranges were 200 to 1500 pg injected mass valeraldehyde equivalent and 180 to 3500 pg acrolein equivalent. Method detection limits (defined as the amount of analyte giving 2 times the background response) for valeraldehyde was 130 pg and for acrolein, 120 pg.

The same column and temperature conditions were used for Hewlett-Packard 5890 capillary GC/^{63}Ni-electron capture detection (ECD) with flows of 5% methane/argon being 3.0±0.4 mL/min, with a detector temperature of 250 °C. The signal was visualized with a Hewlett-Packard 3396 integrator. Linear ranges were also 200 to 1500 pg injected mass for valeraldehyde equivalent, and 180 to 3500 pg for acrolein equivalent injected mass in 2-μL injections. The detection limit for valeraldehyde was 110 pg, and 100 pg for acrolein.

Vapor Generator-Air Dilution System-Exposure Chamber (Figure 2)

Pure air for both the concentrated vapor stream and dilution air branches was generated with a Whatman Zero Air Generator using house compressed air (Figure 2). Extra indicating Drierite and charcoal cannisters needed to be installed after the generator to indicate when the purified air was sufficiently dry and free of organic vapor for use. After 30 min at an input air pressure of 75

pound per square inch, the total hydrocarbon concentration was <0.1 ppm and the RH was <2.0%. The range of the rotameters were: 0.46 to 4.6 L/min for the air dilution branch and 0.1 to 1.5 L/min for the organic vapor concentrated air stream before vapor generation. The air generator was connected to the vapor and water generation sites by 6.4 mm OD stainless steel tubing (type 316). The generators were syringe pumps set at known plunger velocities to generate the desired concentration of organic vapor for dilution, or RH for humidification. Heating tape wrapped around the outside of the stainless steel tubing at the needle exit from the syringe pumps ensured total volatilization of organic vapor or water. The two streams were then routed through a stainless steel T-joint adapter, and the outlet connected by Teflon tubing of 6.4 mm ID secured by butt-to-butt joints with a Tygon collars to a 1-L Greenburg-Smith impinger which acted as a mixing chamber. The entire air generation and dilution system was mounted on a movable two-tiered laboratory cart.

Teflon tubing conveyed the diluted organic vapor into the exposure chamber (Figure 2) through a hole bored on the side near the chamber bottom to just underneath the fan blades, the fan resting at the bottom of the chamber and under the 23-cm diameter ceramic metal plate containing 11 mm diameter holes and a center 30 mm hole. The exposure chamber was a Bel-Art clear polycarbonate vacuum desiccator of height 311 mm, outer diameter 280 mm, flange 273 mm, and supporting plate diameter 230 mm. Remote control with a variac allowed different fan blade velocities and hence face velocities, as well as adequate mixing as shown by direct reading organic vapor analysis and hot wire anemometry. Six samplers were set horizontally on the plate each with a nearby closable hole in the chamber wall for probe insertion for measurement of RH,

temperature, organic vapor concentration, and face velocity. Teflon tubing vented excess vapor to a fume hood. Direct reading confirmation of the diluted organic vapor concentration was only possible for valeraldehyde using a calibrated 11.7 eV HNu PI-101 photoionization detector (PID). The half-times to maximum concentration varied between 5-10 min and the half-times to decrease to baseline varied between 10-15 minutes.

Acrolein concentrations in the exposure chamber were assessed by the published dynamic PFBHA method[7] involving a personal sampling pump set at 10 mL/min, desorption of the O-oxime, and subsequent GC/MS or GC/ECD quantitation using the internal standard method. The whole system was made leakproof through assessment by a soap bubble solution, subsequent appropriate tightening, and/or application of Teflon tape. All plastic tubing was hard-wired with nichrome wire.

Passive Sampler Assembly

PFBHA (300 mg) was dissolved in 25 mL of ASTM Type I water, and the solution added to 2.700 g of Tenax TA in a weighed 250 mL 24/40 ground glass pyrex round bottom. The water was removed by rotary evaporation at 85 °C until a flowing solid occurred. The solid and container were placed in a vacuum desiccator containing indicating Drierite until a constant weight was achieved, usually after three days. A 150.0±1.0 mg weight of coated sorbent was then pelleted by a Parr 2811 bench handpress. The thickness of the pellet was 3.0±0.1 mm and the diameter was 13.0±0.2 mm (Figure 1). The use of methanol solvent instead of water caused formation of a large formaldehyde PFBHA O-oxime background.

For the initial studies a pellet was placed in the Teflon-lined screw cap of dimensions 18 mm OD, 14 mm ID, internal depth 14 mm, and outer height 16 mm. The diffusion path length was 11.0±0.1 mm. In its field form, the screwcap containing the pellet was secured to the bottom inner surface of the outer plastic casing of a 3M 3500 organic vapor monitor[23,24] by duct tape, after removal of the original carbon cloth from the monitor. The Teflon stay of the 3M monitor was cut to allow the pellet to be held securely by one prong to maintain constant diffusion path. The 37-mm silicone membrane was then inserted on the top of the screw cap and then topped by a 37-mm Teflon filter of 10-μm pore size, before being capped with an aluminum seal (Fisher Scientific 06-406-14B) by a crimper (Figure 1). The whole sampler was wrapped in aluminum foil (shiny side out) until sampling.

Methods

Each experiment was done at least in triplicate.

Synthesis of PFBHA O-Oximes

The acrolein and valeraldehyde PFBHA O-oximes were first synthesized by methods already detailed elsewhere[7]. The respective GC/MS purities and yields in % were (97.7±2.3) and (90.0± 2.0) for acrolein, and (99.0±0.9) and (90.6±2.9) for valeraldehyde. The respective GC/ECD purities in % were (98.2±1.7) for acrolein and (96.5±2.5) for valeraldehyde.

Desorption Efficiency of O-Oximes

A volume of 50 μL of hexane solution containing the theoretical amount formed after sampling 8

hours of exposure at the TLV-TWA was spiked onto the pellet. The spiked pellet was held overnight in a desiccator containing Drierite to allow the hexane to dry before desorption with 2.0 mL hexane over 2 hours, 30 sec of ultrasonication, with standing at room temperature over 2 hours before analysis by GC/MS or GC/ECD.

Reaction Efficiency/O-Oxime Recovery for Wet Spiking of Aldehyde and Capacity Studies

A weight of 1.338 mg of liquid valeraldehyde (two times the TLV-8 hour mass) was spiked directly onto the sampling pellet. GC/MS and GC/ECD analyses were then performed as detailed for the desorption of the PFBHA O-oximes. The same procedure was used for acrolein except it was spiked at 1.81 μg (2 TLVx 8 hours) in 50 μL of methanol.

Vapor Exposures

All the exposures in Table 1 were performed at (25±1) °C, (36±2) % RH, and 51 ft/min, the latter being well above the critical face velocity of 15-20 ft/min (7.6 -10.0 cm/s). The total ppm-hr was obtained by summing the area under the ppm versus time exposure plots.PID of acrolein was not possible because of lack of sensitivity. Instead, the dynamic sampling method at 50 mL/min flow rate was utilized simultaneously[7] over the sampling time of the passive sampler.

In a typical run for valeraldehyde (density 0.818 g/mL at 11 °C), the syringe pump was set at 0.200 mL/hr to discharge into a flow rate of 829±17 mL/min which was then diluted to a total flow rate of (2921±44) mL/min for exposure of the passive sampler for 70 min at a theoretical air concentration of 275 ppm or 0.80 the TLV x 8 hour dose. The actual exposure dose as monitored

by PID was about 300 ppm-hours from the area under the ppm versus time curve, or about 0.75 of the TLV x 8 hour dose after correction to 25 °C and 760 mm Hg. The usual protocol involved generating at least 0, 0.25, 0.5, 1.0, 1.5, and 2.0 times the TLV x 8 hour vapor doses over one hour of exposure at temperatures of (9±1) °C, (25±1) °C, and (48±2) °C, and RHs of (3±1)%, (36±2)%, and (79±2)%, and enough concentrations to define the isotherm. The low temperature was achieved by surrounding the exposure chamber inside a styrofoam enclosure filled with ice, and drilled with holes that allowed access to measuring probe holes. The inner chamber temperature was continuously measured with a calibrated thermometer and the RH with a calibrated hygrometer during the exposures. The high temperature was attained by placing the exposure chamber into a Thermolyne Series 5000 Incubator after taking off the glass door. The latter was replaced with same dimensioned plexiglass door (thickness 1 mm) from a print frame drilled with holes to allow access of the measuring probes and power wires. Desorption with 2-mL of hexane followed by GC/MS and/or GC/ECD analysis then occurred.

Capacity Studies

Liquid spiking and aldehyde vapor capacities at (25±1) °C were determined using hexane for desorption, and GC/MS or GC/ECD for analysis.

Adsorption Isotherms

The aldehyde equivalent moles desorbed per unit weight of sorbent q_A was first calculated from the moles of determined O-oxime, correcting for desorption and sampling efficiencies (mole aldehyde/g solid). Other key parameters calculated were: C_A, the concentration (mol/cm^3) of

aldehyde in the air phase corresponding to its air partial pressure p in equilibrium with its O-oxime product on the surface q_A; C_A^*, the molar air concentration in the same units as C_A that corresponds to the air vapor pressure p^* at temperature T in K; and the relative pressure $z=p/p^*= C_A/C_A^*$.

Brunauer-Emmett-Teller (BET) classified equilibrium isotherms for the adsorption of vapors into five major types. A BET isotherm applies for a linear plot of $z/[(1-z)q_A]$ versus z. The slope is $(F-1)/Fn_m$ where n_m is the monolayer coverage in mol O-oxime/g sorbent, and F is a constant related to the activation energy of adsorption E_a. The n_m for a 1:1 stoichiometry at 100% desorption and sampling efficiency is the same as mol aldehyde equivalent reacted/g sorbent. The intercept is $1/Fn_m$. The volume corresponding to n_m is $v_m=1/(\text{slope} + \text{intercept})$. The BET Type I adsorption isotherm is also the Langmuir adsorption isotherm as detected by linearity of a plot of C_A/q_A versus C_A where the slope is $1/n_m$ and the intercept is $1/bn_m$. The constant b is the Langmuir constant and is related to E_a.

If a Freundlich isotherm applies, a plot of ln q_A versus ln C_A is linear with a slope of 1/n (the Freundlich exponential factor, n >1 for the gas/solid phase), and an intercept equal to ln K where K is the Freundlich preexponential factor. If the Dubinin-Radushkevich (D-R) isotherm is descriptive, ln ($q_A = W/V_l$) versus $R^2T^2(\ln^2 z^{-1})$ is linear where W is the observed filled volume of adsorption sites (mL/g), V_l is the liquid molar volume (mL/mole), and R is the gas constant in appropriate units. The D-R slope is $-k''/\gamma^2$, where k'' is a constant (energy^{-2}) related to E_a, and γ

is the affinity coefficient related to the ratio of adsorption potentials in near homologs. The D-R intercept is ln (n_m=W*/V_l), where W* is the limiting maximum volume of the adsorption sites to be filled (mL/g).

Statistics

Analysis of variance type I and II procedures detected significant differences at $\alpha \leq 0.05$ and significant interactions[25]. Means were also compared with Student *t* tests at $p \leq 0.05$[25]. Linear regressions were performed with a SASS statistical package that also provided standard deviations of the slope and intercept as well as correlation coefficients and p-values.

RESULTS

Desorption Efficiencies and Effects of Physical Parameters

The desorption efficiencies for spiked PFBHA oximes of *n*-valeraldehyde and acrolein at 8 x TLV concentration equivalent were 95.6±5.4% and 107.2±5.4%, respectively. The respective recoveries after wet spiking of aldehyde under a number of conditions[14] were 94.0±3.6% and 98.5±7.3%, not significantly different at $p \leq 0.05$. The sampler showed critical face velocities of 7.6-10 cm/s for both aldehydes. The acrolein mass from the theoretical rate of equations (2) and (3) was not significantly different from the mass desorbed, being 96.8±3.4% from 30 experiments at 10 different conditions[14]. This was not so for *n*-valeraldehyde where the average recovery relative to equations (2) and (3) for the same battery of tests was 75.0±1.9%. There were no statistically significant effects of temperature or RH on aldehyde equivalent mass collected at $p \leq 0.05$ in the concentration regions of interest to field passive sampling. The

collected sample was stable (85-110% relative to freshly sampled pellets) for at least 6 months at 4 °C or 25 °C. The sampler shelf life was at least 3 months at 25 °C (84-110% recoveries relative to fresh pellets). The experimental sampling constants k for *n*-valeraldehyde and acrolein were 4.43±0.19 and 7.73±0.57 cm^3/min, respectively. These results are discussed elsewhere[14]. Recoveries for both aldehydes in the dynamic sampling methods based on generated known vapor concentrations were consistently between 80 to 110 % under all conditions of RH and temperature at 50 mL/min[7,14].

Adsorption Isotherms

Figure 3 shows the isotherms of the collection element in terms of moles of aldehyde vapor reacted versus aldehyde moles expected from equation (2).

The vapor and wet spiking capacities were 32.9±3.5 and 28.0±4.8 μmol *n*-valeraldehyde, respectively. The corresponding data for acrolein were 29.1±2.8 and 25.9±5.4 μmol, respectively. There were no statistical differences at $\alpha \leq 0.05$ between vapor recoveries, liquid recoveries, and vapor/liquid recoveries for each aldehyde, or for aldehyde type. The grand average capacity for *n*-valeraldehyde under all conditions was 30.5±3.5 μmol, and that for acrolein was 27.5±2.3 μmol, not statistically different at $p \leq 0.05$. The average limiting capacity is about 29.0 μmol for both, equivalent to a specific limiting capacity of 193 μmol/g sorbent.

For *n*-valeraldehyde, a linear uptake occurred between C_A of 0.011 to 0.055 μmol/cm^3, the latter being equivalent to causing 46% saturation:

$q_A = 1730\ C_A + 0.640 \qquad R^2=0.9876 \qquad p \leq 0.001$ for n=6 (4)

The slope is 1730±31 (μmol/g sorbent)/(μmol/cm^3 vapor), and is related to k in equation (2). The intercept is not significantly different from zero. Therefore this part of the isotherm obeys Henry's Law, and the boundary conditions define the acceptable range for applied passive sampling purposes.

For acrolein over C_A of 0.019 to 0.099 μmol/cm^3 (the latter causing about 95% saturation), the linear regression equation is:

$q_A = 1330C_A + 47.1 \qquad R^2=0.9378 \qquad p \leq 0.05$ for n=4 (5)

This acrolein slope is 1330±170 cm^3/g sorbent and the intercept is nonzero. This slope differs statistically from that for *n*-valeraldehyde at $p \leq 0.05$. Though there is linearity, the large imprecision and the nonzero intercept imply that Henry's Law does not apply over this concentration range. The Henry's Law region for acrolein lies below C_A of 0.019 μmol/cm^3. The pellet specific capacity in μmol/g is not statistically different at $p \leq 0.05$ from that for *n*-valeraldehyde at acrolein C_A values of 0.074 and 0.099 μmol/cm^3, but differ significantly below C_A values of 0.074 μmol/cm^3. Therefore both aldehydes produce O-oximes that are equivalent relative to site occupation at ≥71% of saturation.

The four classical isotherms attempt to describe the approach to saturation. Table 1 compares the parameters from their linearization plots. All the latter were linear for both aldehydes at $p \leq 0.005$ when all data for each aldehyde were considered. Figures 4 and 5 present the Langmuir linearity plots for *n*-valeraldehyde and acrolein respectively, and Figures 6 and 7 show the BET linearity plots for acrolein and *n*-valeraldehyde. Acrolein was best fitted by Langmuir and BET plots at $p \leq 0.001$ with BET being marginally better relative to R^2. *n*-Valeraldehyde adsorption was best characterized by Freundlich and D-R plots, with D-R being the best linearization model relative to R^2. The highest three x-axis points for the Langmuir and BET linearity plots formed a linear subset for *n*-valeraldehyde, these being largely responsible for the linearity of the entire data set for these models, since linearity was not demonstrated without these data. The acrolein BET plot intercept is not statistically different from zero at $\alpha=0.001$, as were the Langmuir and BET intercepts for the three highest concentration data points for *n*-valeraldehyde. All other intercepts differed from zero.

The Langmuir constant b values for *n*-valeraldehyde and acrolein were 2.32±0.72 and 19.8±6.4 cm^3/mol. The BET F factors were 179±14 and 142±13 units, respectively. The respective Freundlich power exponent n values were 1.115±0.046 and 2.32±0.32, and the Freundlich preexponential factors K were 1207±320 and 427±160 units, respectively. The D-R intercepts were both nonzero; both D-R slopes were negative; and intercept and slopes for both aldehydes differed statistically from each other.

DISCUSSION

Three adsorption isotherms contain a sorbent capacity term and an adsorption activation energy term. Table 2 presents these comparisons. The Langmuir or BET I adsorption isotherm is expected to be the most likely adsorption isotherm to describe chemisorption[26]. The Langmuir and BET isotherms are descriptive when intermolecular forces are "vertical" to the surface, not "horizontal", whilst the D-R isotherm does not distinguish the intermolecular forces in the adsorption layer from those in the bulk gas phase. The Freundlich and D-R isotherms occur in situations involving a Gaussian distribution of intermolecular forces, whereas the Langmuir isotherm occurs for situations with nearly monoenergetic "vertical" intermolecular forces.

The theoretical aldehyde capacity for a 150-mg pellet is about 60 μmoles for a compound with one carbonyl group, a theoretical n_m of about 400 μmol/g. The observed average capacity of 29.0±5.8 μmole/150 mg sorbent has an n_m of 193±39 μmol/g. Thus, the theoretical capacity relative to PFBHA is not exceeded, with about half of the total PFBHA molecules used in the chemisorption, with the other half used for nonchemisorptive purposes at experimental saturation. An alternative view is that the adsorbate molecule could occupy or block the occupancy of a second adjacent site, or that the product molecule does so since it cannot be desorbed.

The pellet is porous since both liquid spiking and vapor challenges showed about the same saturation capacity. The corresponding average volume of aldehyde vapor adsorbed is 0.709

cm^3/150 mg or a specific volume of 4.72 cm^3/g, assuming ideality. The corresponding data for a liquid film on 150 mg sorbent for *n*-valeraldehyde and acrolein using their respective measured individual average capacities are 0.00322 and 0.00183 cm^3, with specific liquid volumes of 0.0215 and 0.0122 cm^3/g. No literature X-ray crystallographic data are available for the acrolein and *n*-valeraldehyde O-oximes of PFBHA, and there are no X-ray data for PFBHA.

Adsorption Isotherm Estimations of Specific Adsorption Volume and Area

Table 2 presents the specific capacities calculated from Table 1 for three classical adsorption isotherms. Since all assume liquid film formation on adsorption, the aldehyde liquid film volume equivalents are given also in Table 2.

The Langmuir slope for all the *n*-valeraldehyde data was 0.00122 ± 0.00031 g/μmole. Therefore, n_m is 1/(0.00122±0.00031)= 820±210 μmol/g (Table 2), with poor precision (CV=26%). If only the three highest concentration points are regressed (Range 2 in Table 1; Figure 4), the slope is 0.00431±0.00045 g/μmol, and its n_m is 232±24 μmol/g, not significantly different from the experimental 203±21 μmol/g. The same analysis using BET linearization yielded respective n_m values of 622±100 and 210±16 μmol/g (Figure 7). Neither of these values differed significantly from the corresponding Langmuir n_m at $p \leq 0.05$. For acrolein, there was no significant difference between the Langmuir and BET n_m and there were no linear data subsets (Figures 5 and 6). The D-R analysis produced n_m of 530±36 and 365±52 μmol/g for *n*-valeraldehyde and acrolein, respectively, from the intercepts. The isotherm n_m range between 1.1-4.0 times the average

experimental n_m, and 0.58-2.1 times the theoretical n_m. No isotherm that utilized all the experimental data predicted experimental saturation n_m even for adequate linearization to $p \leq 0.001$, but the BET isotherm came closest (32% overestimate) for acrolein.

While the experimental n_m in terms of μmol/g for either aldehyde are not statistically different (acrolein/*n*-valeraldehyde ratio of about 1.0), this could be explained in at least two ways: common reaction stoichiometry, or common Ideal Gas behavior independent of reaction. If a liquid film forms, the isotherm parameters related to saturation should be related to the experimental liquid aldehyde volume capacity ratio of 0.57±0.10 for acrolein/n-valeraldehyde, or to molecular weight (0.65). Both ratios are surrogates for molecular size. Considering all data for each isotherm, the ratios for the Langmuir, BET, and D-R models for liquid aldehyde capacity are 0.194±0.068, 0.349±0.089, and 0.62±0.13. Only the D-R isotherm ratio is not significantly different from the experimental liquid aldehyde capacity ratio, though its absolute values are not in agreement with experimental ones for the individual aldehydes. When the ratios for capacity in μmol/g are then compared similarly, the respective ratios are 0.31±0.11, 0.389±0.098, and 0.69±0.14. The BET and D-R parameters are self-consistent, but those of the Langmuir isotherm are not. The fact that the D-R n_m are at least twice as large as the experimental saturation data may indicate a multilayer situation in both gas and liquid phases leading to better fitting by the BET and D-R models rather than a monolayer model embodied by the Langmuir isotherm. Alternatively, non-PFBHA parts of the Tenax TA surface may be included in the D-R and BET n_m. Liquid aldehyde phase occurrence before reaction is supported

more any Ideal Gas sole process. though a mixed gaseous-liquid layer model is not eliminated.

A widely used practice for BET II and IV isotherms in computing adsorption site area in multilayer situations is to equate it to the molecular area of the adsorbate as estimated from liquid or solid densities.[26] The assumption is that a condensed phase is the end result of the initial physical adsorption of the aldehyde before actual reaction. For example for liquid *n*-valeraldehyde, the density and molecular weight are 0.81 g/cm^3 and 86.13 g/mol, respectively. There are $0.81/86.13 = 9.4\times10^{-3}$ mol/cm^3 $= 9.4\times10^{-3}\times6.02\times10^{23} = 5.66 \times 10^{21}$ molecules/cm^3. A *n*-valeraldehyde molecule has a liquid volume of 1.77×10^{-22} cm^3, as compared with 1.63×10^{-22} cm^3 for *n*-pentane, 1.70×10^{-22} cm^3 for methyl cyclobutane, 1.67×10^{-22} cm^3 for ethyl cyclopropane, and 2.01×10^{-22} cm^3 for cyclopentane, all calculated similarly for the related hydrocarbon conformers. The addition of the doubly-bonded carbonyl O to *n*-pentane is responsible for an added volume of 0.14×10^{-22} cm^3. If the liquid molecule is also spherical, the minimum diameter of a spherical site that it occupies singly and fully is $2 \times 0.177 \times 10^{-21} \times 21/88 = 0.697$ nm/site, and the equivalent molecular area is $0.349^2 \times 10^{-18} \times 88/7 = 1.53\times10^{-18}$ m^2/site. For the average number of sites per g sorbent that are equivalent to 203 μmol (1.22×10^{20} molecules) of *n*-valeraldehyde, the specific area at experimental saturation is 187 m^2, much larger than is available for uncoated Tenax TA, 35 m^2/g. Not filling PFBHA sites completely will decrease this area, but causes computational problems since distributions have to be assumed that depend on the geometry and disposition of surface PFBHA sites. Similarly, acrolein with liquid density 0.839 g/mL and molecular weight 56.06 g/mole provides the following for the

liquid: molecular volume, 1.10×10^{-22} cm^3; molecular area for a spherical molecule = 1.12×10^{-18} m^2; site spherical diameter= 0.297 nm; and specific surface area = 169±16 m^2/g at 183 μmol/g capacity. It is clear the simple model is not realistic.

Assuming instead that surface-immobilized gas phase *n*-valeraldehyde consists of four equivalent middle right circular cylinders with a frustum of a right circular cone containing the aldehyde group and assuming standard bond lengths and free rotation of isolated σ-bonds, (C=O, 0.122 nm; aldehyde C-H, 0.112 nm; hydrocarbon C-H, 0.1107 nm; C-C, 0.154 nm; non-bonded O, 0.150 nm) and angles (CCO, 123.55°, HCH, 109.3°, HCC, 108.9°), the minimum volume of the immobilized molecule[27] is about 3.0×10^{-23} cm^3, about 5.6 times smaller than a liquid molecule. Gas phase adsorption is still possible according to this model.

The unsaturated part of the acrolein molecule is flat since it is conjugated, but it also has *cis* and *trans* conformers[28]. Regarding acrolein as three frustums of right circular cones and assuming standard bond lengths (C=O, 0.122 nm; aldehyde C-H, 0.112 nm; $C\text{-}C_{aldehyde}$, 0.147 nm; non-aldehyde C-H, 0.109 nm; C=C, 0.134 nm; lone pair O, 0.150 nm) and angles (HCO, 121°; CCO, 123.55°; HCC, 115.5°; HCH, 118°), the minimum volume of the immobilized molecule is 1.73×10^{-23} cm^3, or about 0.58 of that for *n*-valeraldehyde using the same assumptions. This is close to the experimental ratio of liquid aldehyde capacities.

The capacity analysis skews the physical picture towards the extreme where a liquid aldehyde

film before reaction is favored. An activation energy analysis should complement this physical picture if valid.

Estimations of Activation Energy of Adsorption

The forward rate constant k_1 for Langmuir equilibrium related to the number of available surface sites is the reciprocal of the adsorption time which is the average time of stay τ or sticking time of the molecule on the surface.[26] Thus,

$$\tau = \tau_0 e^{Q/RT} \tag{6}$$

where: τ_0 is 10^{-12} to 10^{-13} sec

Q is the energy of adsorption (the interaction energy)

For the reverse process rate constant k_2, if a site is regarded as a two-dimensional potential box, the rate of adsorption will be given by the rate of molecules impinging on the site area σ_0.[26]

$$k_2 = \frac{N_0 \sigma^0}{(2\pi \mathrm{MRT})^{1/2}} \tag{7}$$

Therefore, the Langmuir equilibrium constant b= k_1/k_2 becomes

$$b = \frac{N_0 \sigma^0 \tau_0 e^{Q/RT}}{(2\ \pi\ \mathrm{MRT})^{1/2}} \qquad (8)$$

where

N_0 is the Avogadro's number (6.02×10^{23} molecules/mole)

σ^0 = area of 1 valeraldehyde molecule = 1.52×10^{-14} cm^2

$\tau_0 = 10^{-12}$ sec[26]

Hence, the adsorption energy Q of *n*-valeraldehyde on PFBHA-coated Tenax TA for all its data is 31.2±7.9 kJ/mol, but 40.5±4.2 kJ/mol at the high concentration end. Similarly, acrolein has a Q of 35.9±3.4 kJ/mol. The Q values for the two molecules are not significantly different statistically at $p \leq 0.05$. These results are not in the range of the energies to break chemical bonds, that is >80 kJ/mol. The energies are similar to those of physical adsorption, or hydrogen bonding where energies of 13-32 kJ/mol are known[20]. Furthermore, while the carbonyl group bond length of 0.122 nm is not affected by conjugation in acrolein, its bond energy is raised to 773 kJ/mol, mostly in the π-bond component, due to resonance[20]. This has no effect on Q values, and implies that the π-bond is not implicated in the rate determining step(s).

Since the acrolein data also are fitted by a BET isotherm, the activation energy of adsorption can be calculated also from the BET constant F= 142±13:

$$F = g_0 \exp\left[-(\Delta H_1 - \Delta H_L)/RT\right] \qquad (9)$$

where ΔH_1 is the enthalpy of adsorption of the first layer, and ΔH_L is the heat of liquefaction at temperature T in K, with g_0 being the entropic factor, the latter usually being set equal to 1.0. BET Type II and IV adsorption isotherms occur for $\Delta H_1 > \Delta H_L$, and BET Types III and V for $\Delta H_1 < \Delta H_L$.

For acrolein, ΔH_L is 31.3 kJ/mol using a vapor pressure of pressure at 25 °C of 274 mm Hg and boiling point of 55 °C via the Clausius-Clapeyron equation[29]. Therefore, ΔH_1 is 16.8±40.6 kJ/mol for g_0=1. Similarly, for *n*-valeraldehyde with vapor pressure at 25 °C of 50 mm Hg and boiling point 103 °C[29], ΔH_L is 34.0 kJ/mol, and its ΔH_1 values are 18.8±0.9 kJ/mol using all the data, and 22±78 kJ/mol at the high concentration end. Again, these values are typical of physical adsorption or hydrogen bonding. The ΔH_1 values for both aldehydes are less than the corresponding ΔH_L values at g_0=1, and they do not differ statistically at $p \leq 0.05$ when all data are considered, because of high imprecision. The imprecision may imply that the receptor sites are not monoenergetic relative to adsorption. Thus BET III and V isotherm behavior is predicted. Type V behavior is observed directly for acrolein in the present study

If the Langmuir Q values are used as ΔH_1, the g_0 value for acrolein is 910±2,200, and those for *n*-valeraldehyde are 2.68±0.27 for all the data and 11,000±40,000 for the high concentration data. All the data have high imprecision apart from the *n*-valeraldehyde data that imply g_0 is between 2.4 and 3.0. Assuming g_0 is 2.68, ΔH_1 is 13.9±33.6 kJ/mol for acrolein, 21.8±1.1 kJ/mol for *n*-valeraldehyde for all data, and 25±88 kJ/mol for *n*-valeraldehyde for the high concentration data. Again, Type III or V BET isotherms are expected. Precise entropic data are obtained usually

from temperature dependence of the Arrhenius preexponential factor, but the chemisorption rate is independent of temperature at low surface coverage.

For the D-R adsorption isotherm, the only parameter with energy related units is k" since γ is dimensionless. For the present data, k" $<\gamma^2$ since the D-R slopes are < 1. Traditionally, k" has been associated with the type of adsorption site structure of a sorbent, and has been interpreted to signify a Boltzmann or Gaussian distribution of energy sites, rather than the equal energy sites of the Langmuir or BET I model. Since γ values are not known, k" values cannot be calculated. A process that obeys the D-R adsorption isotherm should have a negligible temperature coefficient. This is so experimentally for the two aldehydes at low surface coverage where passive sampling occurs in the field[14]. This behavior is supportive of a aldehyde vapor phase boundary layer model with liquid film formation just before reaction.

Mechanisms

Microporous Sampler Considerations.

The classical adsorption isotherms were first derived for nonmicroporous surface adsorption. The PFBHA coated pellet of sorbent path width w cm is actually microporous, and the adsorption process may resemble capillary condensation rather than monosurface adsorption. The area of adsorption for microporous sorbents is not defined by sampler dimensions, but rather by micropore number and micropore size distribution. Most modifications of eqn (2) for microporous media leave out the A term and determine A indirectly as done above for the Langmuir, BET, and D-R isotherms.

A common modification of equation (2) for diffusion control uptake into microporous surfaces is [33].

Average flux of A to the sorbent surface Fl_1 in mol cm^{-2} s^{-1} = $(D_A/L)(C_A - C_{surf})$ (10)

where C_{surf} is the concentration of A at the pellet surface.

Average flux of A from the sorbent surface into its interior $Fl_2 = (\zeta \epsilon D_A)\, dC/dx$ (11)

$= (wD_A/\alpha L)dC/dx$ (12)

where ζ is the dimensionless tortuosity factor for interparticle diffusion within the pellet, ϵ is the dimensionless fractional interparticle void volume within the pellet, x is the distance from the pellet surface into the pellet interior where adsorption occurs, and α is the mass transfer resistance, $w/\zeta\epsilon L$.

At steady state:

$$Fl_2/Fl_1 = (\zeta\epsilon L)\,(dC/dx)/(C_A - C_{surf}) = w(dC/dx)/[\alpha(C_A - C_{surf})] \quad (13)$$

If $Fl_1 = Fl_2$

$$\zeta\epsilon L\, dC/dx = (C_A - C_{surf}) = w\,(dC/dx)/\alpha \quad (14)$$

In this chemisorption, C_{surf} is close to zero since the reaction product is formed and retained at the surface.

$$\zeta \epsilon L \, dC/dx = C_A = w(dC/dx)/\alpha \qquad (15)$$

$$dC/C_A = dx/\zeta \epsilon L = \alpha \, dx/w \qquad (16)$$

Integrating between the pellet surface and the average distance x for which vapor penetration occurs from near the surface at which x=0 at concentration C_A,

$$\ln (C_x/C_A) = x/\zeta \epsilon L = \alpha \, x/w = k'x \qquad (17)$$

where x/w is the reduced distance. The w term is usually constant as for the present case, and in pellets that are reproducible x should also be constant.

Thus the constant k', the microporous adsorptivity coefficient in cm^{-1}, is ($1/\zeta \epsilon L = \alpha /w$), and C_x varies exponentially with distance x away from the original exposed surface, usually a micropore.

Mass transfer within the sorbent pellet of thickness w is related to the internal pellet concentration[33]:

$$\zeta \, D \, (d^2C/dx^2) = [(1-\epsilon)K/\epsilon + 1]dC/dt \qquad (18)$$

where concentration equilibrium is assumed between the interparticle void space of macropores, mesopores, and micropores and their adsorbing surfaces with which they are in contact. K is the partition coefficient in mol of A/cm^3 of sorbent per mol of A/cm^3 vapor for the initial physical adsorption assuming a Henry type law. Thus the sampler uptake is the sum of the moles in the air gap, plus those within and on the sampling pellet collected over sampling time t:

$$dn_A/dt = \text{instantaneous uptake} = C_A AD/(L + wn_A/\zeta\epsilon n_{max}) = C_A AD/(L + \propto n_A/Ln_{max}) \qquad (19)$$

From eqn. (18),

$$(dC/dt)/(dn/dt) = 1/dV =$$

$$\zeta C_A \propto^2 (L + \propto n_A/Ln_{max})/(Aw^2[(1-\epsilon)K/\epsilon + 1]) = C_A (L + \propto n_A/Ln_{max})/ (\zeta\epsilon^2 L^2[(1-\epsilon)K/\epsilon + 1]) \quad (20)$$

dV is the volumetric sampling constant of the passive sampler in terms of parameters relating to the microporous nature of the passive sampler. Equation (20) is essentially a first order relationship with a complex first order rate constant. The linearization relationships for the Langmuir and BET I isotherms should therefore be obeyed, and they are for the present system.

For carbonyl compound vapor adsorbing physically to the sorbent[33]:

$$d^2C/dx^2 = 1/(\zeta D_A) [(1-\epsilon)K/\epsilon + 1]dC/dt \qquad (21)$$

$$= (\epsilon\alpha L/wD_A)[(1-\epsilon)K/\epsilon + 1](dC/dt) \quad (22)$$

When $Fl_1 = Fl_2$ from eqn (15),

$$dC/C_A^2 = D_A\, dt/(\zeta\epsilon^2L^2[(1-\epsilon)K/\epsilon + 1]) \quad (23)$$

On integrating this second order kinetic equation from t=0 to time t,

$$1/C_{At} = 1/C_{Ao} - D_A\, t/(\zeta\epsilon^2L^2[(1-\epsilon)K/\epsilon + 1]) \quad (24)$$

The second order rate constant is $D_A/(\zeta\epsilon^2L^2[(1-\epsilon)K/\epsilon + 1])$.

The Freundlich linearization plot produces slopes that could be interpreted as the order of the adsorption process except for the first order case. The slope for *n*-valeraldehyde was 0.897 and 0.433 for acrolein indicative of different rate determining steps. Zero order causes a Henry's Law behavior.

For microporous physical adsorption at low z onto activated carbon diffusive samplers, the D-R adsorption isotherm has been found to predict the same results as does an irreversible adsorption isotherm when K is high for both physical adsorption and chemisorption, or when α is <0.5. This is consistent with the present data obeying D-R linearization plots. Chemisorption that is activated by the collisional energy involved with physical adsorption may also permit high K and

low activation energies. For α >2.0, the design features of the sampler predominate over chemisorptive phenomena.

While the above picture explains why all the linearization procedures for the four classical isotherms are obeyed in the present system, there is still some ambiguity as to why the experimental saturation is half the theoretical, and why the corresponding n_m parameters of the classical isotherms consistently overestimate it. According to the microporosity model, this could be caused by either $\zeta\epsilon$ in equation (11) being 0.5, α in equation (12) being a factor with w/L= 0.273±0.012, K in equation (18) being 0.5, or a mixture of some variation of all of these. Since the whole pellet is microporous, $\zeta\epsilon$ is more likely to be 0.5 because of tortuosity and restrictions within the pellet inner air spaces so that α is 0.546 with a minimum CV of 4.4% assuming w and L are constant. Therefore, the microporous adsorptivity coefficient in equation (17) would be about 0.182 cm^{-1}. In practice, K must be constant to have a practical passive sampler, but for the present sampler this is so only below 71% of experimental saturation as shown by the Henry's Law analysis. Above this, both aldehydes have the same K, which progressively decreases for both in the same manner as saturation is approached. While *n*-valeraldehyde showed linearity between 0.011-0.055 μmol/cm^3 and obeyed Henry's law, acrolein showed linearity between 0.019-0.099 μmol/cm^3 but did not obey Henry's Law. The *n*-valeraldehyde/acrolein sampling constant ratio was[14] 0.573±0.067(CV=12%), whereas the slope ratio of the Henry's Law plots was 1.30±0.19 (CV=15%). Thus K for *n*-valeraldehyde is 1.0 but is probably 0.573 for acrolein between 0.019-0.099 μmol/cm^3 compared with 1.0 in the Henry's Law region below 0.019 μmol/cm^3. Thus while *n*-valeraldehyde chemisorption depends only on

$\zeta\epsilon$ in its Henry's Law region, acrolein chemisorption depends also on K in the linear concentration range studied for its isotherm.

The Addition Reaction Mechanism.

Aldehydes can donate electrons but not protons. PFBHA like hydroxylamines can form a lattice of strong hydrogen bonds and therefore can donate protons to a suitable acceptor molecule like an aldehyde. The various tetrahedral intermediates during carbonyl addition have weak hydrogen bonding as does the product oxime[32].

Having PFBHA sites in close proximity may facilitate proton donation, and efficient reaction due to the orienting effect of the surface PFBHA molecules. The fluorinated benzyl group of PFBHA surface coating is probably held flat on the flat aromatic rings of the Tenax TA surface, with the -oxyamino hydrochloride end sticking out from the surface and being able to rotate propeller-like. The bond lengths expected are: C=C, 0.139 nm; C-F, 0.135 nm; ring C-sidechain C, 0.150 nm; C-H, 0.111 nm; C-O, 0.141 nm; O-N, 0.124 nm; and N-H, 0.1034 nm. Side chain rotation could occur about its C-O, O-N, and N-H σ-bonds. The electronegative ring fluorines may enhance the inductive effect of the sidechain oxygen lone pairs, thus strengthening the C-O and O-N σ-bonds relative to isolated ones and weakening the N-H bonds. Thus the PFBHA sites could be already oriented for optimum reaction, and ready to donate at least one proton. The chloride ion of about 0.167 nm radius has to be replaced by the lone pairs of the incoming oxygen of the carbonyl atom. The chemisorption reaction must be through the carbonyl end of the aldehyde. The minimum volume of the end carbonyl group and α-carbon group is 1.23-1.37

x 10^{-23} cm^3. In the most stable tetrahedral transition state intermediate, the bond lengths are: C-O, 0.1427 nm; O-H, 0.0956 nm; C-H, 0.1096 nm; C-C, 0.154 nm; C-N, 0.147 nm. The various angles are now HCO 108.9°, HCH 123.55°, and CCH 109.5°. In the O-oxime product, the N=C bond length is about 0.128 nm in length.

There are at least two very different possible mechanisms: the initial physical adsorption to Tenax TA is rate determining, followed by an acid catalyzed chemisorption process as in equation (1); or the initial physical adsorption process is not rate-determing and one or two proton transfers are rate determining before or after the addition reaction. The transfers could occur through hydrogen-bonding, expected to be in the range of 13-42 kJ/mol. The smallest ΔH_1 values are those calculated from BET F values, and if they represent one proton activation, the Langmuir Q values probably could imply two such transfers, or one such transfer plus another process like physical adsorption of aldehyde molecules.

Since much of the Tenax TA surface is not covered, there is a large potential for physical adsorption. Acrolein and long chain aldehydes are known to be retained by uncoated Tenax TA[16-18]. Such a physical adsorption mechanism involving an extrinsic precursor would allow lateral diffusion on the surface to increase the opportunity to find a free site. A possibility is that the first layer consists of valeraldehyde molecules lying flat across the surface allowing reaction with PFBHA sites to cause the chain to lift from the surface. When the surface gets crowded at about 71% of experimental saturation, a lyotropic phase change may occur so that the hydrocarbon tails become vertical to allow more adsorption of aldehyde molecules on the surface

sites. This may simulate multilayer adsorption at high surface coverages, but a Henry's law behavior exists at low C_A values where passive sampling actually occurs. This adsorption mechanism may also involve an extrinsic precursor[30,31] postulated to involve at least two PFBHA surface sites to facilitate a preferred orientation for reaction to account for the low activation energy, and the postulated proton transfers or assisted physical adsorption. This mechanism complements the model that invokes K and $\zeta\epsilon$ to explain the observed phenomena.

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Table 1. Comparison of Adsorption Isotherm Linearity Plots

Aldehyde	Isotherm	Range	Linear Plot Regression Parameters Slope	Intercept	R^2	p
n-Valeraldehyde	Langmuir	1	122(31) x 10^{-5}	5.25(0.29) x 10^{-4}	0.8314	<0.001
		2	431(45) x 10^{-5}	43(69) x 10^{-6}	0.9947	<0.05
		3	33(25) x10^{-5}	56(244) x 10^{-5}	0.0479	0.856
	BET	1	165(27) x 10^{-5}	1935(94) x 10^{-7}	0.8441	<0.001
		2	479(36) x 10^{-5}	59(209) x 10^{-7}	0.9943	<0.05
		3	56(27) x 10^{-5}	21(111) x 10^{-5}	0.1228	0.483
	Freundlich	1	0.897(0.037)	7.096(0.115)	0.9881	<0.001
	D-R	1	-0.2914(0.0098)	6.273(0.068)	0.9921	<0.001
Acrolein	Langmuir	1	399(38) x 10^{-5}	201(46) x 10^{-6}	0.9658	<0.001
	BET	1	417(38) x 10^{-5}	29(70) x 10^{-6}	0.9671	<0.001
	Freundlich	1	0.433(0.060)	6.056(0.164)	0.9306	<0.005
	D-R	1	-0.120(0.016)	5.90(0.14)	0.9343	<0.005

BET, Brunauer-Emmett-Teller; D-R, Dubinin-Radushkevich; Range 1, all data; Range 2, highest three data for x-axis; Range 3, all data except range 2 data; Slope and Intercept data are in appropriate units and provided as arithmetic mean and standard deviation; R^2, square of the correlation coefficient; p, statistical probability of non-linearity using Student *t*.

Langmuir Adsorption Isotherm: $C_A/q_A = C_A/n_m + 1/(bn_m)$ where C_A is the concentration of adsorbate in the vapor phase (mol/cm^3), q_A is the moles adsorbed/g sorbent, n_m is the mole of adsorbate/g for single occupation of all sites, and b is the Langmuir constant (cm^3/mol)

BET Adsorption Isotherm: $z/(1-z)q_A = (F-1)z/Fn_m + 1/Fn_m$ where z is the reduced pressure, and F is the BET constant

Freundlich Adsorption Isotherm: $\ln q_A = \ln K + 1/n \ln C_A$ where K is the Freundlich preexponential factor, and n is the Freundlich exponential factor.

Dubinin-Radushkevich Adsorption Isotherm: $\ln q_A = \ln n_m - (k"R^2T^2 \ln^2 z^{-1})/\gamma^2$ where $n_m = W^*/V_l$ where W* is the limiting potential specific volume (mL/g) at temperature T in K, V_l is the liquid molar volume (mL/mol), k" is a constant (energy^{-2}), γ is the affinity coefficient, and R is the gas constant in appropriate units.

Table 2. Comparison of Sorbent Capacity and Activation Energy Terms for Three Adsorption Isotherms for *n*-Valeraldehyde and Acrolein. Terms are Defined in Table 1.

Aldehyde	Isotherm	Range	Capacity (μmol/g)	Liquid Aldehyde Capacity (μL/g)	Energy (kJ/mol)
n-Valeraldehyde	Langmuir	1	820(210)*	86(22)*	31.2(7.9)
		2	232(24)	24.4(2.5)	40.5(4.2)
	BET	1	622(100)*	46.1(7.4)*	28.8(1.4)
		2	210(16)	22.1(1.7)	17(62)
	D-R	1	530(36)*	39.1(2.7)*	
	Experimental		203(21)	21.5(2.2)	
Acrolein	Langmuir	1	251(24)*	16.7(1.6)*	35.9(3.4)
	BET	1	242(22)*	16.1(1.5)*	19(46)
	D-R	1	365(52)*	24.3(3.5)*	
	Experimental		183(15)	12.2(1.0)	

*, Significantly different from experimental value at Student *t* at $p \leq 0.05$

LEGENDS FOR FIGURES

Figure 1. Cross Section of the Passive Sampler
Figure 2. Aldehyde Vapor Generation and Exposure Chamber System
Figure 3. Vapor Isotherm for Acrolein and *n*-Valeraldehyde in Terms of Aldehyde Reacted Versus Theoretical Aldehyde Reacted
Figure 4. Langmuir Linearization Plot for *n*-Valeraldehyde
Figure 5. Langmuir Linearization Plot for Acrolein
Figure 6. BET Linearization Plot for Acrolein
Figure 7. BET Linearization Plot for *n*-Valeraldehyde

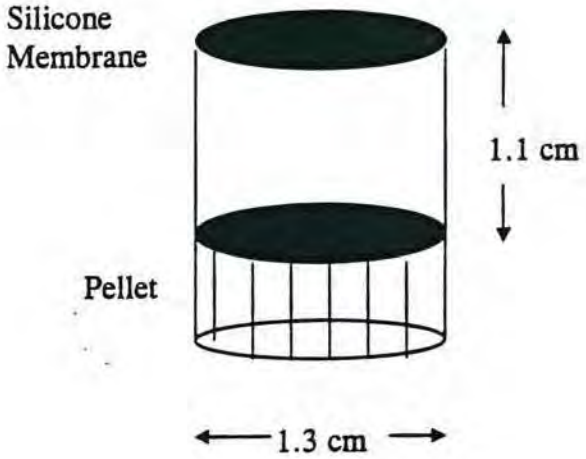

Fig. 1.

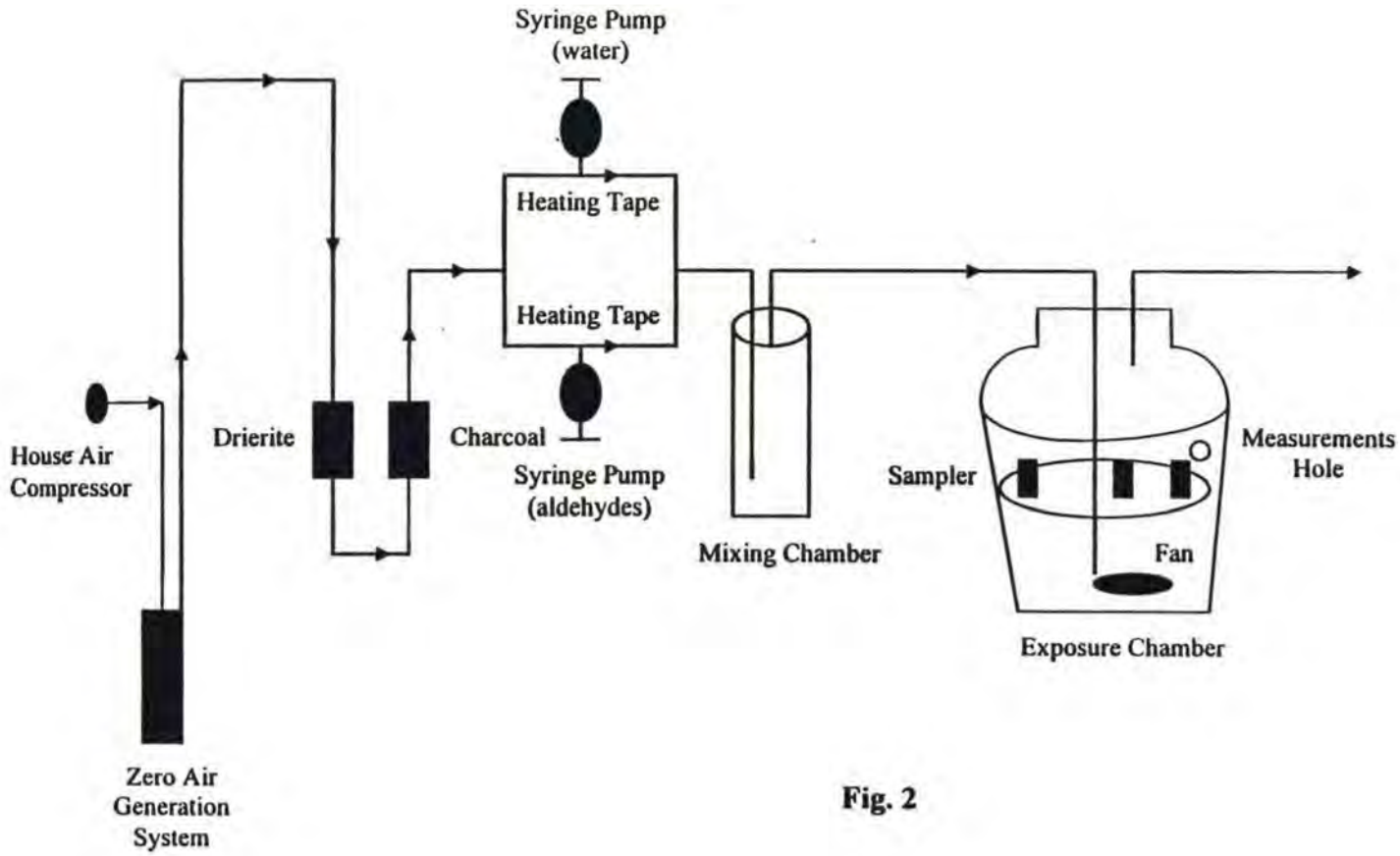
Syringe Pump
(water)
Heating Tape
Heating Tape
House Air
Compressor
Drierite
Charcoal
Syringe Pump
(aldehydes)
Mixing Chamber
Sampler
Measurements
Hole
Fan
Exposure Chamber
Zero Air
Generation
System

Fig. 2

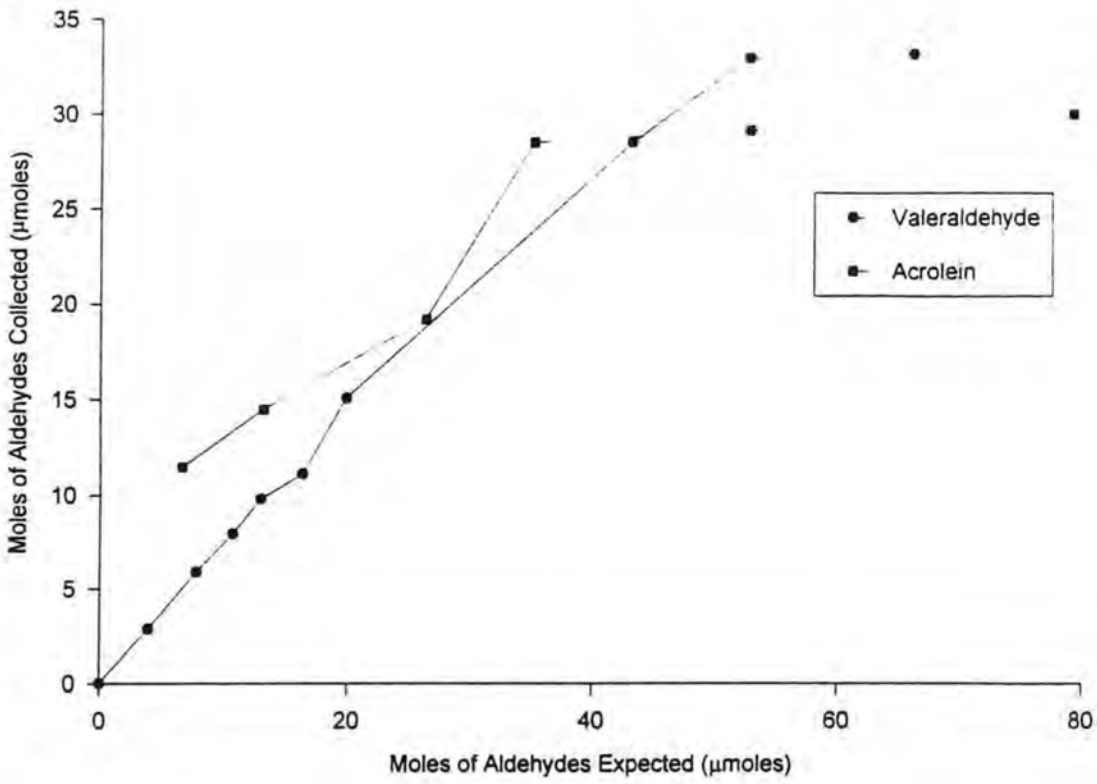
35
30
25
20
15
10
5
0
0
20
40
60
80
Moles of Aldehydes Collected (µmoles)
Moles of Aldehydes Expected (µmoles)
Valeraldehyde
Acrolein

Fig. 3.

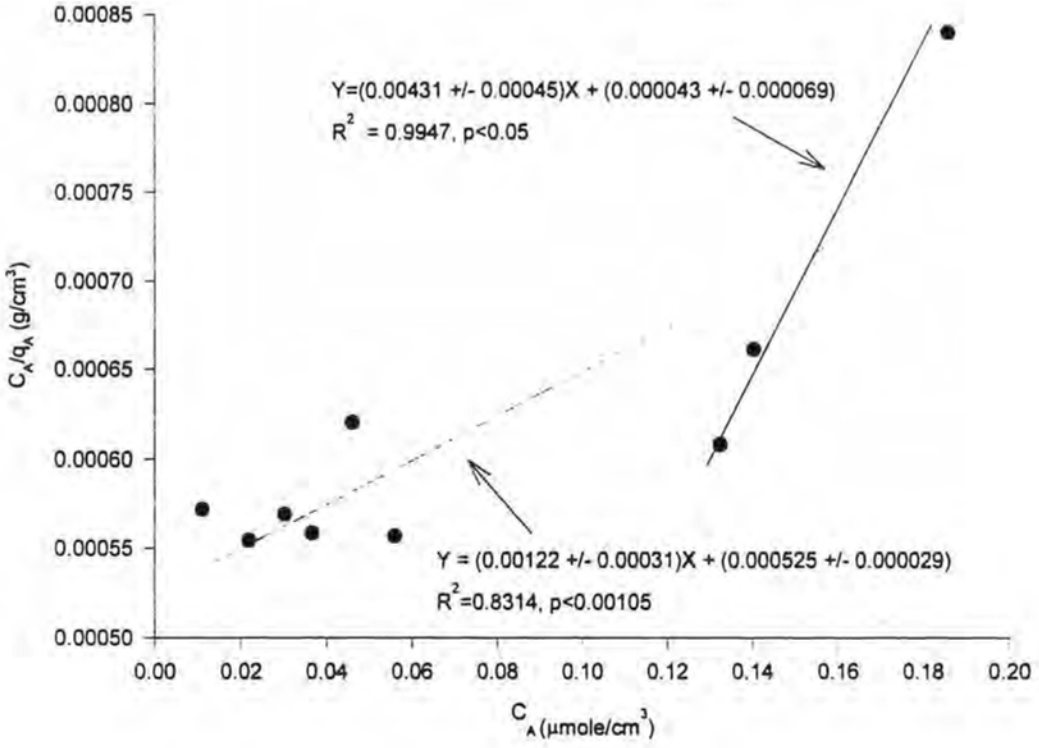
0.00085
0.00080
0.00075
0.00070
0.00065
0.00060
0.00055
0.00050
C_A/q_A (g/cm^3)
Y=(0.00431 +/- 0.00045)X + (0.000043 +/- 0.000069)
R^2 = 0.9947, p<0.05
Y = (0.00122 +/- 0.00031)X + (0.000525 +/- 0.000029)
R^2=0.8314, p<0.00105
0.00
0.02
0.04
0.06
0.08
0.10
0.12
0.14
0.16
0.18
0.20
C_A (µmole/cm^3)

Fig. 4

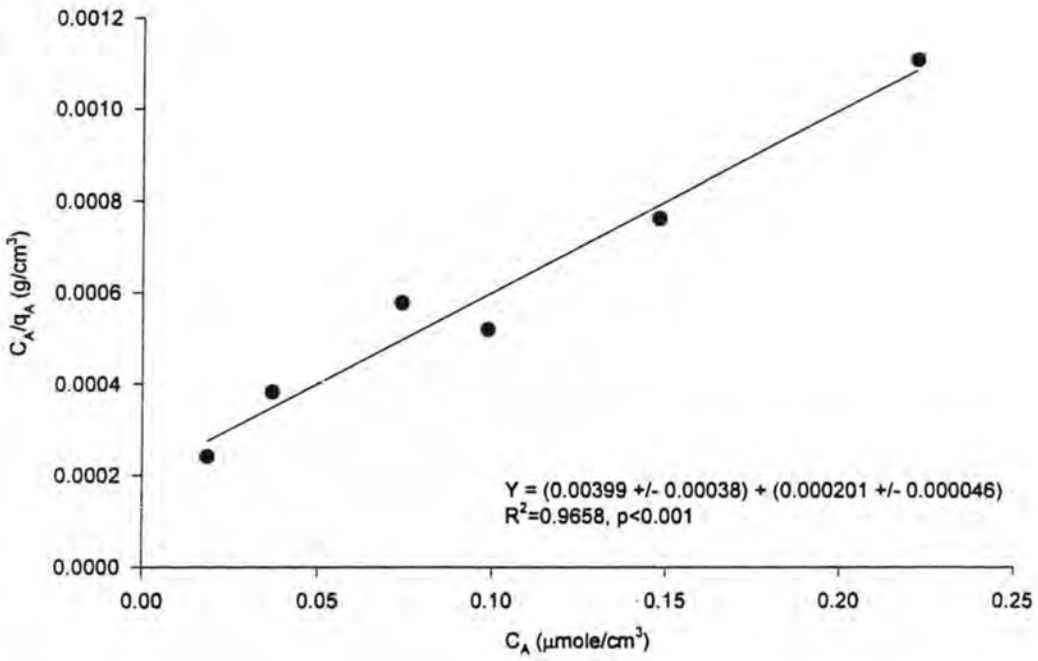
0.0012
0.0010
0.0008
0.0006
0.0004
0.0002
0.0000
C_A/q_A (g/cm^3)
0.00
0.05
0.10
0.15
0.20
0.25
C_A (μmole/cm^3)
Y = (0.00399 +/- 0.00038) + (0.000201 +/- 0.000046)
R^2=0.9658, p<0.001

Fig. 5

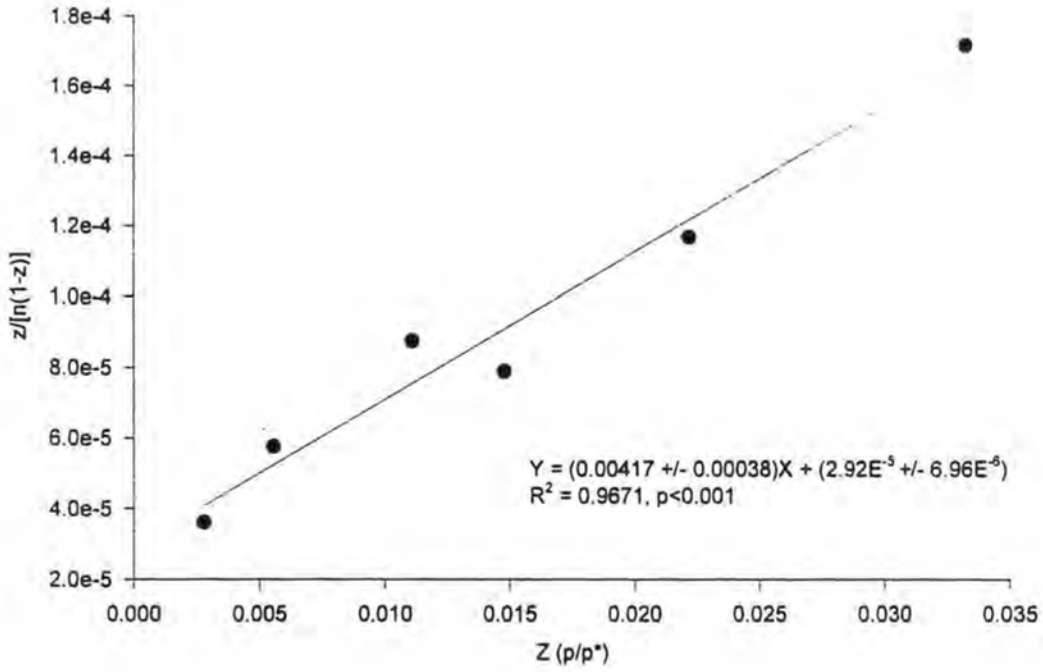
1.8e-4
1.6e-4
1.4e-4
1.2e-4
1.0e-4
8.0e-5
6.0e-5
4.0e-5
2.0e-5
z/[n(1-z)]
0.000
0.005
0.010
0.015
0.020
0.025
0.030
0.035
Z (p/p*)
Y = (0.00417 +/- 0.00038)X + (2.92E-5 +/- 6.96E-6)
R2 = 0.9671, p<0.001

Fig. 6

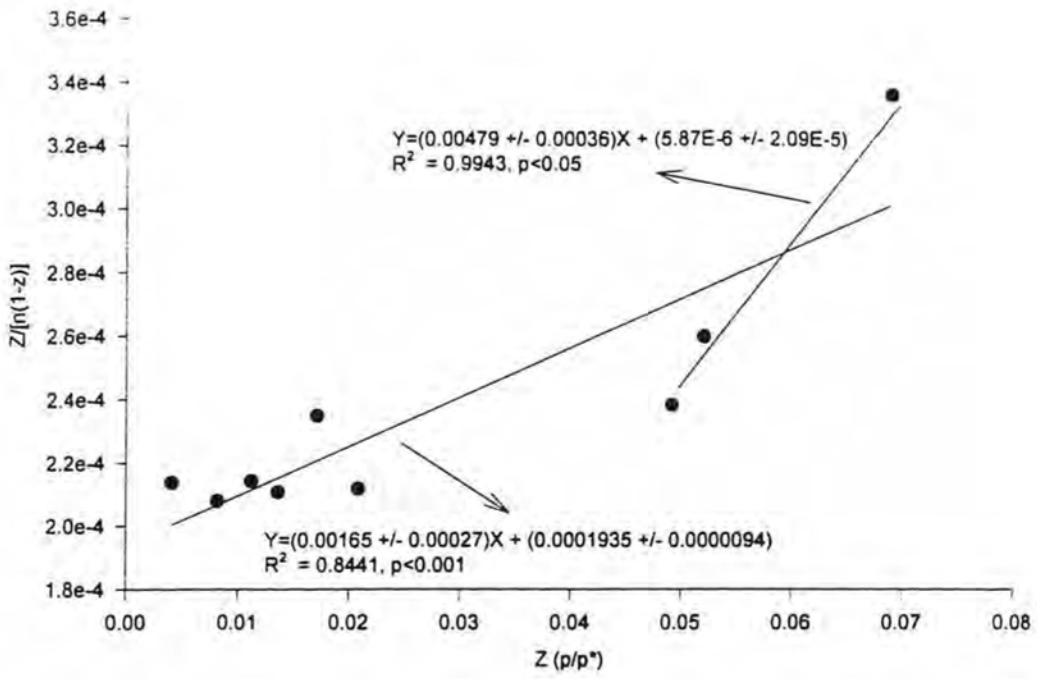
3.6e-4
3.4e-4
3.2e-4
3.0e-4
2.8e-4
2.6e-4
2.4e-4
2.2e-4
2.0e-4
1.8e-4
Z/[n(1-z)]
Y=(0.00479 +/- 0.00036)X + (5.87E-6 +/- 2.09E-5)
R2 = 0.9943, p<0.05
Y=(0.00165 +/- 0.00027)X + (0.0001935 +/- 0.0000094)
R2 = 0.8441, p<0.001
0.00
0.01
0.02
0.03
0.04
0.05
0.06
0.07
0.08
Z (p/p°)

Fig. 7

APPENDIX 8

ALDEHYDE AND KETONE AIR EXPOSURES

FOR UNIVERSITY WORKERS

Shane S. Que Hee[a*], Shih-Wei Tsai[a], Yang Shen[a],

Joseph Raab[b] and Victor Kennedy[c]

[a] Department of Environmental Health Sciences and UCLA Center for Occupational and Environmental Health, School of Public Health, University of California, 10833 Le Conte Avenue, Los Angeles, California 90095-1772

[b] Office of Environment, Health and Safety, University of California,10920 Wilshire Blvd Suite 1850, Los Angeles, California 90024-1604

[c] Medical Center Building and Safety, University of California, 10833 Le Conte Avenue, Los Angeles, California 90095-6941

* Corresponding author

ABSTRACT

The aim of the study was to measure formaldehyde, glutaraldehyde and acetone personal breathing zone concentrations by dynamic and passive solid sorbent sampling based on O-(2,3,4,5,6-pentafluoro-benzyl)hydroxylamine hydrochloride coated Tenax TA. The job practices investigated in a University job setting included pouring operations for dispensing and waste combination purposes, routine tissue fixing, and solvent cleaning purposes. A 11.7 eV photoionization detector and a dual flame ionization/10.6 eV photoionization instrument provided a collaborative analysis, in the "sniffing" mode. Acetone personal air concentrations were validated by parallel sampling with a charcoal tube, in addition to direct reading instruments calibrated to be direct reading to acetone. The workplace protection factors against formaldehyde and acetone for personnel who wore respirators are reported for the first time using passive samplers. Industrial hygienists who use direct reading instruments to monitor prolonged pouring and combination of formalin and glutaraldehyde for workers should also wear the appropriate respirator protection.

Introduction

Universities are now major employers of industrial hygienists because of the diversity of occupational health hazards that students, staff, and faculty may face during research, service, and teaching activities. While academia train and educate industrial hygienists for the job market, the institutions of higher learning also need to act as models relative to their occupational health and safety programs and management. Many such as the Office of Environment, Health and Safety at the University of California in Los Angeles provide comprehensive protection programs for employees (UCLA Office of Environment, Health and Safety, 1997). Major issues such as accidents and accident prevention, safety procedures, emergency responses, fire and explosion issues, sewer and stormwater issues, evacuation procedures, asbestos, blood-borne pathogens and safety associated with microorganisms, radiation hazards, employee training, animal care hazards, and human subject hazards are considered. The major elements of the industrial hygiene subprogram at the UCLA campus include personal monitoring and environmental testing, personal protective equipment, hazard communication, ergonomics, project design, laser safety, hearing conservation, health inspections and training, and non-ionizing radiation. The environmental health subprogram covers food facility inspections and food handler training, cooling tower water testing, potable water quality, wastewater, storm water and surface water protection, toxic air emissions, and swimming pool sanitation. The Hazardous Materials Management subprogram oversees chemical transportation, storage, and inventory, material safety data sheets (HMDSs), chemical waste management, hazardous material spill response, pollution prevention, waste minimization, and compliance testing. Often universities separate the health and safety responsibilities for university hospitals and health science centers

because of administrative and occupational reasons. Thus dermatitis, blood- and breath- borne pathogens, use of chemosterilants and drugs, sharps safety, biomedical waste disposal, physician, nurse, dentist, pharmacy, dental staff, operating room, and pathology and clinical laboratory concerns are focussed on (Sarri et al 1991; Arrington and McDiarmid, 1993; Christman and Gandsman 1994; Haddock et al 1994; Rattner et al. 1994; Ulin 1997; Wood 1997; Wall et al. 1997; Turner et al. 1999; Wright et al, 1999). There can be still much overlap, however, when the responsibilities are split.

The traditional training of an industrial hygienist is perfectly suited to combatting the hazards of university workplaces when the hazards are known, as for example, when accurate HMDSs are available, or if noise, ionizing radiation, and nonionizing light sources are present. If industrial hygiene training programs have good air quality, indoor air, water quality, and analytical chemistry courses or areas of focus, the industrial hygiene graduate can also cope when chemical hazards are not known, a common occurrence in campus offices, waste containment yards, laboratories, and in the ambient environment.

Thus for air sampling for gases and vapors , the traditional strength of industrial hygiene chemical sampling, the Columbia University School of Public Health staff investigated university air quality and ergonomic issues in 1985 (Stellman et al. (1985)). University art department facilities have been shown to produce adverse exposures to methyl cellosolve acetate and toluene (Lucas and Salisbury, 1992). High ethylene oxide exposures have been detected for personnel at a University hospital (Sobaszek et al. 1999). Exposure to anesthetics has been

evaluated in a German university hospital (Byhahn et al. 1999).

The gas/vapor exposure of concern for the present study on workers at the University of California at Los Angeles is to aldehydes and ketones. The major aldehydes whose personal breathing zone air exposures have been reported in hospital and embalming environments are formaldehyde and glutaraldehyde (Kerfoot and Mooney 1975; Williams et al. 1984; Moore and Ogrodnik 1986; Norback 1988; Holness and Nethercott 1989; Binding and Witting 1990; Stewart et al 1992; Leinster et al 1994; Korczynski 1994; Naidu et al 1995; Bennett et al. 1996; Tharr 1996; Pisaniello et al. 1997; Niven 1997; Wellons et al 1998). There have been no reports of exposures at Universities. Formaldehyde has a Occupational Safety and Health Administration (OSHA) permissible exposure limit (PEL) of 0.75 ppm with a Short Term Exposure Limit (STEL) of 2 ppm; there is an American Conference of Governmental Industrial Hygienists (ACGIH) 1999 Ceiling Limit of 0.3 ppm; and the National Institute for Occupational Safety and Health (NIOSH) Recommended Exposure Limit (REL) is 0.016 ppm, and a Ceiling limit of 0.1 ppm. Formaldehyde is usually used as a 37-41% (w/w) aqueous concentrate containing 6-12 % (w/w) methanol stabilizer or as more dilute aqueous solutions, for its embalming and tissue preservative uses. It has been the major tissue fixative since 1893. Area concentrations of up to 18 ppm (22 mg/m^3) have been reported. Glutaraldehyde has no OSHA regulation; the ACGIH 1999 Ceiling Limit is 0.05 ppm with a sensitizer notation; and the NIOSH Ceiling is 0.2 ppm. Glutaraldehyde is usually available as Cidex aqueous solutions for chemosterilant and for embalming. It has been used since 1955. Air concentrations of up to 1.4 ppm (5.7 mg/m^3) have been reported.

Another carbonyl compound that is much used in chemistry and medical laboratories as a solvent but especially for cleaning purposes is acetone. The OSHA PEL is 1,000 ppm; the NIOSH REL is 250 ppm; and the 1999 ACGIH TLV is 500 ppm and the STEL is 750 ppm.

The present study evaluates some formaldehyde, glutaraldehyde, and acetone exposures to staff at the University of California at Los Angeles.

Methods

Some pouring operations were monitored by direct reading instruments calibrated with 100 ppm toluene for formaldehyde and glutaraldehyde monitoring or 100 ppm acetone for acetone monitoring: a 11.7 eV H Nu 101 photoionization (PID) detector; and the Foxboro dual PID/flame ionization detector (FID) Model TLV1000A set to read and accumulate data every 15 seconds. In some cases, a explosimeter calibrated with n-pentane, the Micromax LEL meter, was also utilized.

Unit Processes Evaluated.

Formalin Dispensing and Waste Combination. One male worker (Worker 1 in Table 1) wore a full-face negative pressure respirator with a purple color acid gas/formaldehyde canister that was quantitatively fitted as well as latex gloves, paper shoe covers, and a laboratory coat. He also wore prescription spectacles without side shields. 10%-Formalin was dispensed from the stop cock of a carboy inside the fume hood. After the formalin was used in paraffin block-making machines, spent liquid was poured manually into a polyethylene carboy and the carboy

capped. Several carboys containing waste were then transported by cart to a distant waste disposal/dispenser room. The formalin was combined in a 55-gallon drum using a grounded funnel, and each emptied carboy was refilled with fresh 10% formalin, recapped, and replaced onto the cart for transport back to the histology laboratory. Respirator protection was always used for this step. The disposal room was ventilated, but the door was always left open nevertheless. The combination of wastes took about 60 min. The laboratory produced about 50 gallons of formalin waste daily. Filled 55-gallon drums were picked up by campus Environmental Health and Safety personnel at a nearby loading dock for transport to the waste containerization yard.

Workers Who Used Formalin for Tissue Specimen Preservation. The male worker (Worker 2A in Table 1) wore both a dynamic and passive sampler as he uncovered lids, and dipped specimens. He wore a laboratory coat, apron, and latex gloves. Both 10% and 37% formalin are used in the procedures. Small containers holding the specimens are brought into the laboratory for processing. The specimen is removed and placed in a plastic cassette about two-thirds filled with formalin. The lid is then closed, and the sample shaken gently. The specimen may be dissected with a scalpel on a cutting board, and portions placed into other cassettes. Two adjacent workstations were equipped with a local exhaust slot hood (Shandon Upshaw) of about 23 cm x 117 cm. Sinks were on either side of the hoods, and were used to dispose of spent formalin solution in the cassettes. Stores of fresh formalin were situated above the sinks. Each specimen was recorded manually into a log sheet and the diagnosis and description dictated into a microphone actuated by a foot control.

A female worker (Worker 2B in Table 1) dissected tissue samples immersed in 10% formalin. Controls included local exhaust ventilation (fume hood), and a laboratory coat, and latex examination gloves were worn.

Combining Aldehydes at the Hazardous Waste Containerization Facility. This facility received campus-wise hazardous wastes to store and bulk within 90 days after generation of the waste. The aldehyde wastes in small and large containers were combined at this facility into large 55-gallon drums. The three male workers (Workers 3A, 3B, and 3C in Table 1) who were involved in the bulking procedure were experienced in hazardous waste procedures. They wore quantitatively fitted full-face negative pressure respirators with combination organic and acid gas cartridges, as well as Tyvek coveralls, steel-toed shoes covered with protective Tyvek booties, double gloves (nitrile inner/neoprene outer, both duct-taped just above the wrist), and hard hats. The workers positioned their bodies upwind during the pouring. All metal funnels utilized were electrically grounded to a chain-link fence. About 60 gallons of aldehydes are combined in one week. A recording industrial hygienist (Worker 3D in Table 1), who also wore a quantitatively fitted negative pressure respirator, also wielded direct reading instruments during the pouring.

Acetone Exposures. Dispensing acetone from a electrically-grounded 55-gallon drum on a loading dock into 1-gallon containers for laboratory use required two people. One (Worker 4A in Table 1) operated the hand pump by turning the handle a defined number of times while the other (Worker 4B in Table 1) placed the funnel and pump hose in the container to be filled and indicated when the container was almost full, with no spills. The pumper was about 1 m from

the emission source, whereas the hose/funnel attendant was about 40 cm from the same source. The monitoring industrial hygienist (Worker 4C in Table 1) was about 2 m downwind from the emission source. A volume of 29 gallons was dispensed. The task is done once a week over a normal time span of about 2 hours. The hose/funnel attendant also had acetone exposures monitored with a charcoal tube solid sorbent, using NIOSH Method 1300. None of the workers wore respirators, and they were dressed in laboratory coats, disposable gloves, and safety glasses.

A laboratory assistant (Worker 5 in Table 1) who wore a disposable charcoal-lined paper respirator mask (MSA Affinity Organic Vapor), a laboratory coat, and safety glasses with side shields whilst cleaning equipment with acetone from a squeeze bottle was then evaluated by passive sampling inside and outside the mask.

Carbonyl Compound Sampling and Analysis Method. The carbonyl compound air sampling techniques were dynamic sampling and passive sampling. Passive sampling was used whenever a disposable or negative pressure respirator was worn so as to obtain lapel and inside the respirator concentrations. If a dynamic sampler was also present, the passive sampler was placed on the opposite lapel.

The dynamic sampling technique utilized portable personal pumps set at 25 to 50 mL/min for Tenax TA solid sorbent coated with 20% (O-2,3,4,5,6-pentafluorobenzyl)hydroxylamine hydrochloride (w/w) in glass tubes (200 mg/100 mg; each 7 cm x 5 mm ID) as described elsewhere (Wu and Que Hee, 1997; Shen and Que Hee, 1999; Lin and Que Hee, 1999). The

calibrated SKC pocket pump was clipped inside the worker's laboratory coat pocket, the tygon tubing connected to the sampler clipped behind the shoulder, and the sampler then clipped downward at the lapel. The pumps were calibrated before and after sampling. The average flow rate was calculated if the two readings did not differ by more than 25%.

The passive sampler was a 13-mm diameter pellet of 10% (w/w) PFBHA on Tenax TA contained in the plastic shell of a 3M model 3500 passive sampler, as described elsewhere (Tsai and Que Hee, 1999a,b,c). A clip allowed the passive sampler to be affixed hanging down to the lapel in the breathing zone after the protective aluminum foil was removed, ensuring the membrane side faced outward. A piece of duct tape facilitated the insertion of the passive sampler inside the respirator so that it did not impede vision or swing loose. Aluminum foil covered the passive samplers inside 3 M 3500 cans before and after exposure for transport and storage. Aluminum foil was also used to cover the dynamic samplers after capping with tube caps and replaced into its screw-capped glass tube transport container.

In the laboratory, the passive samplers were desorbed with 2.0 mL of hexane for 2 hours, and the dynamic samplers were desorbed with 3 mL of hexane for 2 min whilst ultrasonicating, as described elsewhere. Aliquots were both analyzed by capillary gas chromatography (GC) using either a ^{63}Ni-electron capture detector (ECD) from Hewlett Packard at 250 °C for 95% argon-methane carrier at 3 mL/min, or by selected ion mass spectrometry (SIM-MS) at m/z 181 using a helium carrier flow rate of 3.0 ml/min. The capillary column for both detectors was a 30 m and 0.32 mm inner diameter fused quartz column, chemically bonded with a 1-μm DB-1701 film.

The 70 eV electron impact positive ion quadrupole mass spectrometer was a Hewlett Packard Model 5988A with transfer line at 250 °C and ion source at 260 °C. The gas chromatograph in both cases was a Hewlett Packard Model 5890 with the injector held at 250 °C. The aldehydes and ketones were first subjected to a mass spectrometer solvent delay of 0.75 min at 105 °C, or for GC/ECD an initial 0.5 min wait at 105 °C. Thereafter both column ovens were subjected to a temperature ramp of 10 °C/min up to 220 °C, holding at 220 °C for 10 min thereafter. The GC/ECD output was displayed on a Hewlett Packard Model 3396 reporting integrator.

The internal standard method using pure O-oxime standards synthesized by methods described elsewhere (Wu and Que Hee, 1995; Tsai and Que Hee, 1999a,b,c; Shen and Que Hee, 1999; Lin and Que Hee, 1999; and Wiesenthal and Que Hee, 1999) and decafluorobiphenyl was used for quantifications over the range 0.80 ng to 80 ng. Apart from formaldehyde, both *E*- and *Z*-isomer peak areas were used for quantitation purposes.

The O-oxime content was converted to aldehyde equivalent and divided by the air volume sampled to determine the air concentration, corrected to 25 °C and 760 mm Hg. The sampling constants for the passive sampler were from Tsai and Que Hee, (1999b) for the aldehydes, or from the Fuller-Schettler-Giddings (Tsai and Que Hee, 1999a) method for acetone.

Results and Discussion

The results of all the personal breathing zone sampling measurements and supporting direct reading instrument readings are shown in Table 1. These results provide the first reported

workplace protection factors (WPFs) measured for respirators worn for protection against exposures to aldehydes and ketones. In addition, adjunct direct reading instrument air sampling was conducted to demonstrate the reasonableness of the passive and dynamic integrated personal breathing zone air sampling concentrations.

Formaldehyde Exposures. In general, the passive sampling concentrations are higher than dynamic sampling results. This was so for Workers 1b, 2A,3Aa, 3B, and 3C for whom the ratio was generally between 3.6 to 14, although one value of 86 was obtained for Worker 3Aa. The reasons for this may be related to different exposure situations for the opposite lapels since both techniques provide identical results for the same generated atmospheres when sampled continuously (Tsai and Que Hee, 1999a,b,c; Lin and Que Hee 1999; Shen and Que Hee, 1999). In addition, the sampling surface cross-sectional areas may make a difference in the sampling of intermittent vapor bands relative to continuous exposures. Thus if a sampling cross section is of importance for the 13 mm diameter pellet and the 5 mm ID dynamic sampler, the factor difference expected is $(6.5/2.5)^2 = 2.6^2=6.8$ in favor of the passive sampler. However, diffusive flow rates are lower than for dynamic samplers, a fact that allows dynamic samplers to sample more contaminated air when both samplers are exposed to a constant concentration of contaminant or when exposure periods are long for intermittent exposures. However, if there is a sharp vapor front, the dynamic sampler will dilute its sample with uncontaminated air whereas a larger passive sampler must be exposed to a higher instantaneous mass from the same vapor or gas front assuming it is homogeneous in concentration. This bears investigation in chamber experiments.

In situations where a passive sampler was placed both inside and outside a respirator for the same sampling time so making moot the precise value of the sampling constant and making sampling cross-sectional areas uniform, the WPFs for the full-face negative pressure respirators were: Worker 1b, 5.0; Worker 3Ab, 3.1; Worker 3B, >4; and Worker 3C, 140. Of these only the respirator for Worker 3C definitely performed adequately, since there was an inside concentration lower than the NIOSH REL of 0.016 ppm, the rest generally having WPFs lower than 10. The NIOSH WPF guideline for full facepiece negative pressure respirators is 50, and with a quantitative fit factor of 500 (42 CFR 84). The respirator for Worker 3B may have been adequate. Clearly, WPFs should be measured directly, and passive samplers are ideal since they do not import air through the face-respirator seal or the cartridge as dynamic samplers may. The independence of these passive and dynamic samplers to the effects of relative humidity and temperature are also advantages (Wu and Que Hee, 1995; Tsai and Que Hee, 1999a,b,c; Shen and Que Hee, 1999). There is no doubt that the pouring process at the hazardous waste yard and for the pathology laboratory required respirator protection since Worker 1a was exposed to 0.96 ppm, Worker 1b to 0.65 ppm, Worker 3A was exposed to 5 and 19 ppm, and Worker 3C to 1.4 ppm, all being near or above the OSHA guideline of 0.75 ppm, assuming these exposure conditions continue for 8 hr. However only the respirator for Worker 3C afforded adequate protection. All passive sampler concentrations for the inside of respirators exceeded the ACGIH Ceiling Limit of 0.3 ppm, except for Worker 1b, 3B, and 3C. The hygienist monitoring the hazardous waste yard exposure using direct reading instruments (Worker 3D) also required improved respirator protection from formaldehyde exposure since the inner concentration of 0.17 ppm exceeded the NIOSH Ceiling Limit of 0.1 ppm though not the ACGIH Ceiling Limit of 0.3

ppm. The personal breathing zone air concentration measured outside the respirator for Worker 3Ab is among the highest such concentration so far reported.

The direct reading instrument readings at the hazardous waste yard were also the highest. Gas bag calibration of the 11.7 eV PID showed the following response factors for the following 100 ppm vapors relative to toluene as 100%: 25% aqueous glutaraldehyde, 14%; 38% formalin containing 10% methanol, 24%; 10% methanol, 8%; valeraldehyde, 48%. Formaldehyde actually had a response factor of 16% after accounting for methanol presence.

Glutaraldehyde. Worker 3Aa was also exposed to glutaraldehyde as well as to formalin (Table 1). The passive sampler air concentration was 0.26 ppm, above the ACGIH Ceiling Limit of 0.05 ppm and the NIOSH Ceiling of 0.2 ppm. The 11.7 eV PID recorded concentrations up to 7 ppm toluene equivalents in the breathing zone. Thus respirator use is still required when combining glutaraldhyde.

Acetone. None of the personal breathing zone samples for Workers 4A, 4B , and 4C (recording industrial hygienist) exceeded any guidelines for acetone. In spite of a little breakthrough, the dynamic charcoal tube concentration agreed well with that obtained with the PFBHA passive sampler for Worker 4B. The TVA 1000 direct reading instrument in both its FID and PID modes was also in substantial agreement with the integrated samplers. However, explosive atmospheres were generated near the container opening. Use of a funnel generated much lower concentrations than simply putting the hose end in the container opening. Probably the vortex

effect and closure of the container opening by the discharging funnel prevented gushing of vaporised material out of the container, and this practice is recommended to minimize exposures. Workers should stay upwind as much as possible and not be in a confined space situation.

The laboratory assistant (Worker 5 in Table 1) had very low exposures to acetone. The low WPF of 1.4 did not matter. Use of a squeeze bottle where controlled streams of acetone were produced for cleaning purposes did not cause adverse acetone exposures.

Conclusions

Prolonged pouring operations of formalin and glutaraldehyde require the use of full-facepiece respirator protection whose WPF is best ascertained by passive samplers rather than by dynamic samplers which may disrupt respirator protectiveness. Such pouring operations should be conducted with the operators upwind to the funnel on a concrete loading dock or floor. If pouring occurs in a room, the latter should have its own exhaust ventilation system. Industrial hygienists who monitor breathing zone air concentrations of these chemicals should protect themselves with a quantitatively-fitted respirator, whose WPF should be found.

Acknowledgments

NIOSH RO1 OH 03120 and the UCLA Center for Occupational and Environmental Health were the funding sources. The following EHS252G students are thanked: Bart Ashley; Kristine Bell; Ted Benchoff; Mary Black; Marlene Chuek; Pat Harris; Barbara Kim; Soo Young Kim; Tony Lee; David Leigh; Kyle Lim; Lori Maeda; Keummi Park; Bill Peck; Gerald Pineda; Sara Richards; Peter Sarmicanic; Peng-Cheng Sung; Karina Tatyan; and Jenny Wang.

Table 1. Air concentrations. sampling times and direct reading instrument information for various sampling situations for formaldehyde, glutaraldehyde, and acetone.

Aldehyde	Worker		Flow Rate/ Time Sampled ($mLmin^{-1}$/min)	TWA (ppm)	Direct Reading Instrument Data (PID in ppm Toluene equivalents) and other Comments
Formaldehyde	1 a	PO	9.97/77	0.96	PID: 0-30 ppm in open door of pouring
		PI	9.97/32	0.74	room; 0-15 ppm in laboratory
	b.	PO	9.97/61	0.65	Pouring room only; WPF is 5.0
		PI	9.97/61	0.13	
		D	26/61	0.18	
	2A	P	9.97/111	0.54	
		D	54.5/111	0.080	
	2B	P	9.97/185	0.16	
	3Aa	P	9.97/35	4.9	PID: Average 87 ppm inside funnel
		D	43/33	0.057	during pouring; 7 ppm near breathing zone; 300 ppm at drum exhaust
	3Ab	PO	9.97/12	19	PID: 10-150 ppm about the breathing
		PI	9.97/11	6.1	zone with geometric mean 28 ppm over 5 min; WPF is 3.1
	3B	PO	9.97/25	0.30	WPF >4;

		PI	9.97/30	<0.07	
		D	54/12	0.03	
	3C	PO	9.97/18	1.4	WPF, 140; PID, 1-30 ppm with TWA
		PI	9.97/22	0.01	11±8 (34 readings) in bz
		D	54/12	0.10	TLV-1000 FID: 50-3000 ppm with
					TWA 860±750 (32 readings) in bz
	3D	PI	9.97/16	0.17	
Glutaraldehyde	3Aa	P	4.46/35	0.26	PID: up to 7 ppm in bz; 240 ppm near
					drum opening
Acetone	4A	P	7.47/37	77	
		D	46.9/37	27	
	4B	P	7.47/39	73	TVA 1000 PID bz STEL: 79 ppm;
					bz TWA, 58 ppm; up to 773 ppm
		D	CT 59/39	89	TVA 1000 FID bz STEL: 98 ppm;
					Up to 1990 ppm
					PID: 0-500 ppm in bz; TWA, 69 ppm
					Exposimeter often exceeded 10% (v/v)
					pentane alarm value at funnel
	4C	P	7.47/37	33	
	5	PO	7.47/60	1.1	The disposable respirator had a

PI	7.47/60	0.80	WPF of 1.4

Worker 1, Male formalin dispenser and combiner; Worker 2A: Male pathology technician; Worker 2B: Female pathology technician; Worker 3A, male hazardous waste yard pourer; Worker 3B, male hazardous waste yard pourer; Worker 3C, male hazardous waste yard pourer; Worker 3D, industrial hygienist wearing quantitatively fitted negative pressure respirator; Worker 4A, male acetone pumper; Worker 4B, male acetone funnel/hose attendant; Worker 4C, male industrial hygienist; Worker 5, male acetone user wearing charcoal-lined paper disposable respirator

Lower case letters after the worker number signify a different sampling day.

P, passive sampler; PI, passive sampler inside respirator; PO, passive sampler outside respirator; D, dynamic sampler; CT, charcoal tube; TWA, time weighted average concentration; PID: 11.7 eV HNu 101; WPF, workplace protection factor; TVA 1000 PID, Foxboro TLV 1000 10.6 eV photoionization detector reading in real ppm; TVA 1000 FID, Foxboro TLV 1000 flame ionization detector reading in real ppm; bz: breathing zone

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