

## A Simple and Highly Sensitive Colorimetric Detection Method for Gaseous Formaldehyde

Liang Feng,<sup>†,‡</sup> Christopher J. Musto,<sup>†</sup> and Kenneth S. Suslick<sup>\*,†</sup>

Department of Chemistry, University of Illinois at Urbana-Champaign, 600 South Mathews Avenue, Urbana, Illinois 61801, and Department of Instrumentation and Analytical Chemistry, Dalian Institute of Chemical Physics, Chinese Academy of Sciences, 457 Zhongshan Road, Dalian 116023, P. R. China

Received December 8, 2009; E-mail: ksuslick@illinois.edu

Formaldehyde is widely used in the manufacture of many resins, polymers, and construction materials. The thermal or chemical decomposition of such materials, however, occurs surprisingly readily from urea–formaldehyde foam insulation, particle board, and formaldehyde-based resins.<sup>1</sup> Formaldehyde is a probable human carcinogen, an allergen, and an intense irritant of eyes and mucous membranes and is therefore highly problematic as an indoor pollutant.<sup>1–3</sup> The ability to detect formaldehyde at very low concentrations is critical: the World Health Organization has set a safe-exposure standard of 80 ppb averaged over 30 min;<sup>2</sup> for comparison, OSHA has set the permissible exposure limit (PEL) at 750 ppb and the immediately dangerous to life or health (IDLH) limit at 20 ppm.<sup>3</sup>

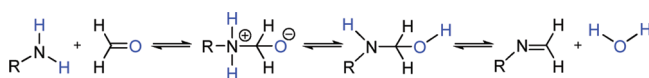
While there are numerous methods for detecting and measuring gaseous formaldehyde, there remains a need for an inexpensive, sensitive, and rapid analytical technology. Past analytical approaches include relatively expensive instrumentation<sup>4</sup> (e.g., electrochemical, gas chromatographic, optical, chemiluminescent, etc.) but also inexpensive colorimetric methods, which see some field use.<sup>5</sup> A number of colorimetric or fluorometric methods for the detection of formaldehyde have been proposed<sup>5</sup> (e.g., hydrazones, triazoles, pararosaniline, etc.). These prior methods are generally relatively slow (typically >30 min), often cumbersome and multistep, and frequently lack sensitivity. Thus, there remains a pressing need for the development of a rapid, sensitive, and highly convenient formaldehyde detection method. Herein, we report a simple and highly sensitive colorimetric method for fast formaldehyde detection.

Most colorimetric formaldehyde detection methods are based on nucleophilic addition of an amine to the aldehyde, forming an imine via a carbinolamine intermediate, as shown in Scheme 1. For an appropriate choice of the primary amine (e.g., one having delocalized conjugated substituents), the imine formation generates a color change, so great effort has gone into the synthesis of sophisticated self-indicating primary amines.<sup>5</sup>

In contrast, we have found that it is better and easier to use a simple pH indicator to detect the change in basicity upon the reaction of a nonvolatile primary amine with formaldehyde. An amine-terminated polymer was used to create a reactive matrix for formaldehyde detection. In order to screen for the optimal choices of indicator and polymeric amine, an array of six different pH indicators in five different polymer formulations [see Table S1 in the Supporting Information (SI)] was printed onto poly(vinylidene difluoride) (PVDF) membranes.

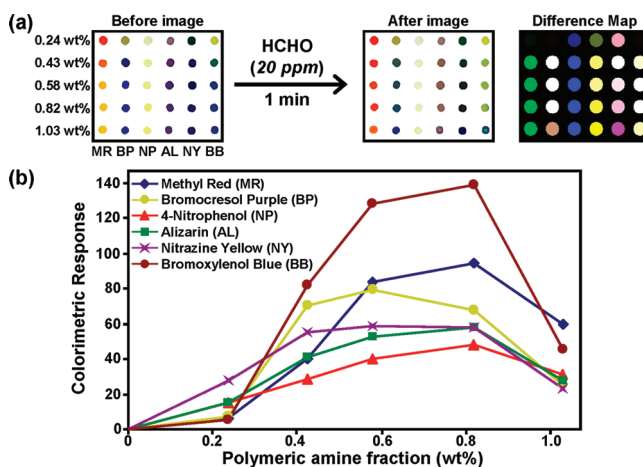
The inexpensive and disposable array was imaged using an ordinary flatbed scanner (Figure S1 in the SI), as described elsewhere for other colorimetric sensor arrays.<sup>6</sup> For each spot in

**Scheme 1.** Nucleophilic Addition of a Primary Amine to Formaldehyde, Forming an Imine



the array, the red, green, and blue values were measured before and after exposure to gaseous formaldehyde, and color difference maps were generated. As shown in Figure 1a, after 1 min of exposure of 20 ppm (IDLH) formaldehyde, obvious color changes are readily observed even by eye. Simply subtracting the original control image from the sample image provides a color-change profile. Figure 1b shows the titration curve of six different pH indicators doped into a poly(ethylene glycol) (PEG) polymer blended with a small amount of the same PEG with amine termination after exposure to 20 ppm formaldehyde for 1 min. Amine–PEG concentrations of ~0.7 wt % gave the largest overall color changes. The response of these polymer films to formaldehyde is very rapid: as shown in Figure S2, the response to 20 ppm formaldehyde is generally >90% complete in less than 1 min.

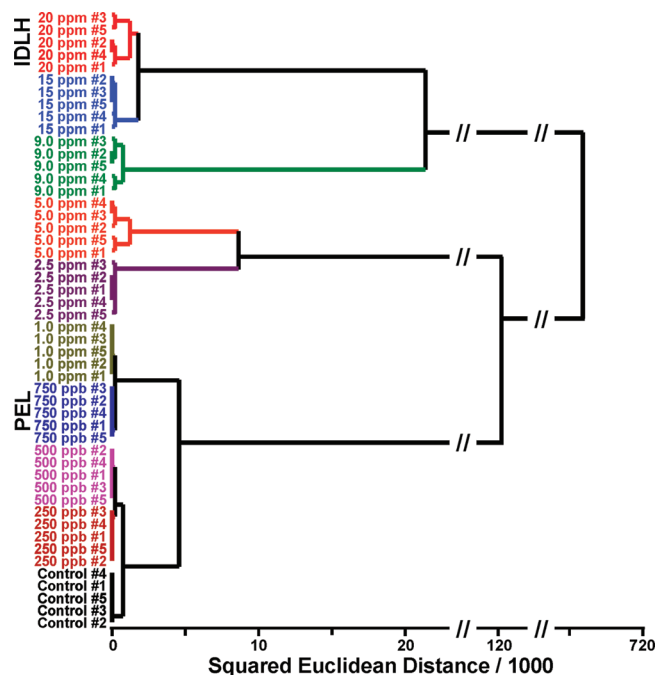
The colorimetric response of these polymeric films can be used for quantitative analysis of formaldehyde concentrations. After 1 min of exposure to formaldehyde at different concentrations, the array shows different color change patterns that are easily distinguished by eye even without statistical analysis (Figure S3). For quantitative comparisons, a simple statistical approach, hierarchical cluster analysis (HCA),<sup>7</sup> was used. HCA is a model-free classifica-



**Figure 1.** (a) Image of the array before and after 1 min of exposure (500 scmm) to 20 ppm formaldehyde at 298 K and 50% RH, along with the color-change profile. Different polymeric amine weight percentages are listed for six pH indicators. (b) Colorimetric response [i.e.,  $(\Delta R^2 + \Delta G^2 + \Delta B^2)^{1/2}$ ] of each pH indicator versus polymeric amine wt % after 1 min of exposure to 20 ppm formaldehyde; indicator abbreviations are also shown.

<sup>†</sup> University of Illinois at Urbana-Champaign.

<sup>‡</sup> Chinese Academy of Sciences.



**Figure 2.** Hierarchical cluster analysis (HCA) for formaldehyde at different concentrations. All of the experiments were run in quintuplicate; no confusions or errors in classification were observed in 50 trials, as shown.

tion scheme based on the grouping of the analyte vectors (i.e., changes in RGB values) by Euclidean distances in their full dimensionality. Figure 2 shows the HCA dendrogram for 1 min exposures to gaseous formaldehyde concentrations ranging from 20 ppm (IDLH) down to 250 ppb (i.e., one-third of PEL). Remarkably, in quintuplicate trials, all of the concentrations showed tight clustering with no errors or misclassifications out of 50 cases. One may also base a quantitative analysis simply on the total response of the array (i.e., total Euclidean distance of changes in RGB), as shown in Figure S4. By increasing the exposure time of the array, we were able to quantify formaldehyde concentrations as low as 50 ppb. Color difference maps and the HCA dendrogram (Figure S5) show that 50, 75, and 100 ppb as well as a control were accurately identified after 10 min of exposure.

For many prior formaldehyde detection techniques, errors stemming from changes in relative humidity (RH) and/or temperature have proven to be highly problematic in real-world applications. For this reason, we selected hydrophobic colorants and an extremely hydrophobic PVDF membrane to minimize the effects of changing humidity. The array itself is unaffected by changes in RH from 10 to 90% relative to 50% RH, as shown in Figure S6. Humidity effects during formaldehyde detection were also evaluated (Figure S7) and found to be only minor even at high humidity. Temperature changes (from 4 to 50 °C) did not significantly affect the overall response (Figure S8).

Acetaldehyde, butyraldehyde, and benzaldehyde were also tested to evaluate the selectivity of our method. None of these aldehydes showed any significant response at ~15 ppm even after 5 min of exposure (Figure S9). The high selectivity stems from decreased charge (and hence diminished electrophilicity) of the carbonyl carbon in substituted aldehydes in comparison with formaldehyde. Even acetaldehyde showed only a very weak response at high concentrations (>200 ppm).

To discriminate formaldehyde from potential acidic or basic interferent vapors, we added a row of several pH indicators in neutral

or base-treated forms (Table S2). These added indicators permit discrimination of formaldehyde from other acidic or basic interferents (e.g., SO<sub>2</sub>, NO<sub>2</sub>, NH<sub>3</sub>, etc.). While this does not permit facile identification of formaldehyde in the presence of such interferents, it does prevent false positives, since formaldehyde itself does not trigger a response from the nonamine polymer films of pH indicators. This array was extensively tested against 10 common interferents: second-hand smoke, diesel fuel exhaust, gasoline exhaust, floor stripper, Windex, Fantastik, and Clorox bleach at 2% of their saturation vapor concentrations; SO<sub>2</sub> and NO<sub>2</sub> at their respective PEL concentrations; and CO<sub>2</sub> at ~375 ppm (i.e., the ambient concentration) (Figure S10). Only Windex and SO<sub>2</sub> provided significant interference, but the additional indicators prevented false positives. The shelf life of the printed arrays is excellent, with constant response for more than 1 month after printing. Using these arrays as a screening tool, we generated a minimalist colorimetric sensor for formaldehyde detection consisting of three spots: BB in a 0.6 wt % amine-PEG film for formaldehyde detection and BB and basic NY in a nonamine PEG film as false-positive discriminators against acidic and basic interferents, respectively.

In summary, we have developed a quite simple and highly sensitive colorimetric detection method for gaseous formaldehyde. Using ordinary pH indicators in an amine-functionalized polymer film, one can discriminate formaldehyde concentrations over a wide range: from 20 ppm (IDLH) down to 250 ppb within 1 min and down to 50 ppb (7% of the PEL) within 10 min. These color changes are easily observed even by eye (Figure S11). We have recently constructed and are in the process of testing a fully functional prototype handheld device that makes use of an inexpensive white LED and an ordinary CMOS camera.<sup>6</sup> Combined with a low-dead-volume cartridge, this handheld device provides a rapid, highly sensitive, and quantitative method for the portable monitoring of formaldehyde at various concentrations.

**Acknowledgment.** This work was supported through the NIH Genes, Environment and Health Initiative (U01ES016011).

**Supporting Information Available:** Experimental procedures, additional figures, and database. This material is available free of charge via the Internet at <http://pubs.acs.org>.

## References

- (1) Leikauf, G. D. In *Environmental Toxicants: Human Exposures and Their Health Effects*, 3rd ed.; Lippmann, M., Ed.; Wiley: Hoboken, NJ, 2009; pp 257–316.
- (2) *WHO Air Quality Guidelines for Europe*, 2nd ed.; World Health Organization: Copenhagen, 2000.
- (3) (a) Armour, S. J. *International Task Force 40: Toxic Industrial Chemicals (TICs)—Operational and Medical Concerns*. U.S. Government Printing Office: Washington, DC, 2001. (b) <http://www.cdc.gov/niosh/hpg/npgd0293.html>.
- (4) (a) Jaffrezic-Renault, N.; Dzyadevych, S. V. *Sensors* **2008**, *8*, 2569. (b) Ferus, M.; Cihelka, J.; Civiš, S. *Chem. Listy* **2008**, *102*, 417. (c) Pal, R.; Kim, K. H. *J. Sep. Sci.* **2007**, *30*, 2708.
- (5) (a) Mohr, G. J. *Anal. Bioanal. Chem.* **2006**, *386*, 1201. (b) Toda, K.; Yoshioka, K. I.; Mori, K.; Hirata, S. *Anal. Chim. Acta* **2005**, *531*, 41. (c) Kawamura, K.; Kerman, K.; Fujihara, M.; Nagatani, N.; Hashiba, T.; Tamiya, E. *Sens. Actuators, B* **2005**, *105*, 495. (d) Gibson, L. T.; Kerr, W. J.; Nordon, A.; Reglinski, J.; Robertson, C.; Turnbull, L.; Watt, C. M.; Cheung, A.; Johnstone, W. *Anal. Chim. Acta* **2008**, *623*, 109. (e) Maruo, Y. Y.; Nakamura, J.; Uchiyama, M. *Talanta* **2008**, *74*, 1141. (f) Suzuki, Y.; Nakano, N.; Suzuki, K. *Environ. Sci. Technol.* **2003**, *37*, 5695. (g) Mohr, G. J.; Spichiger, U. E.; Jona, W.; Langhals, H. *Anal. Chem.* **2000**, *72*, 1084.
- (6) (a) Rakow, N. A.; Suslick, K. S. *Nature* **2000**, *406*, 710. (b) Suslick, K. S. *MRS Bull.* **2004**, *29*, 720. (c) Suslick, K. S.; Bailey, D. P.; Ingison, C. K.; Janzen, M.; Kosal, M. A.; McNamara, W. B., III; Rakow, N. A.; Sen, A.; Weaver, J. J.; Wilson, J. B.; Zhang, C.; Nakagaki, S. *Quim. Nova* **2007**, *30*, 677. (d) Musto, C. J.; Lim, S. H.; Suslick, K. S. *Anal. Chem.* **2009**, *81*, 6526. (e) Lim, S. H.; Feng, L.; Kemling, J. W.; Musto, C. J.; Suslick, K. S. *Nat. Chem.* **2009**, *1*, 562.
- (7) (a) Scott, S. M.; James, D.; Ali, Z. *Microchim. Acta* **2007**, *156*, 183. (b) Johnson, R. A.; Wichern, D. W. *Applied Multivariate Statistical Analysis*, 6th ed.; Prentice Hall: Upper Saddle River, NJ, 2007.

JA910366P