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Fabrication of Plastic Microfluid Channels by Imprinting Methods

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Microfluidic devices have been fabricated on poly(methyl methacrylate) substrates by two independent imprinting techniques. First-generation devices were fabricated using a small-diameter wire to create an impression in plastics softened by low-temperature heating. The resulting devices are limited to only simple linear channel designs but are readily produced at low cost. Second-generation devices with more complex microchannel arrangements were fabricated by imprinting the plastic substrates using an inverse three-dimensional image of the device micromachined on a silicon wafer. This micromachined template may be used repeatedly to generate devices reproducibly. Fluorescent analtyes were used to demonstrate reproducible electrophoretic injections. An immunoassay was also performed in an imprinted device as a demonstration of future applications.

In the past few years there has been an explosion of interest in the field now generally known as microfluidics.[1–7] An important challenge in developing the first generation of microanalytical systems is in the successful design and fabrication of the microcapillary channels. These channels will ultimately be incorporated into self-contained analytical systems with detection and processing elements and will be used to perform chemical or biochemical measurements. Fluid transport in the microchannels may be accomplished using electroosmotic,[8–10] thermal,[11,12] or mechanical pumping;[13] and chemical selectivity may be achieved using electrophoresis or by chromatographic principles such as adsorption, affinity, or ion exchange.

Most prototype devices to date have been fabricated in glass or silica- or silicon-based substrates. Photolithographic processing techniques are used to produce channels in the surface of a planar substrate that are then covered with a plate of similar material.[14] An advantage of using these materials is that their electrophoretic and chromatographic properties[15] and surface derivatization chemistries[16] are extensively characterized in many cases. Recently, a wet-chemical etching protocol for fast-etching glasses (such as soda lime glass) has been reported that substantially decreases the surface roughness typically associated with channels produced in these substrates.[17] Materials that etch more slowly, including quartz, are generally used to create channels with a smoother appearance but require highly resistant masks made of metal or polysilicon. The mask deposition step raises the cost of fabricating channels in these substrates. Channels etched into silicon crystals using similar techniques, although with different chemical etchants, have very smooth and reproducible features.[13,18]

Microfluid channels etched into glass and quartz can be subsequently sealed by high-temperature annealing. The sealing process is not trivial, and most researchers report low device yields when closing channels by this method. Anodic bonding is a simple procedure that may be used to seal hybrid quartz–silicon devices.[18,19] Devices prepared entirely in silicon may be sealed by wafer bonding.[20] Although successful prototype devices have been prepared using silica- or silicon-based substrates, the associated fabrication techniques, including channel fabrication and sealing, are generally difficult and/or expensive to implement.

Polymer substrates are promising alternatives for microfluid devices. Industrial interest in utilizing plastics for the production of microanalytical systems is primarily driven by the fact that these materials are less expensive and easier to manipulate than silica-based substrates. Plastics lend themselves readily to casting, molding,[21] laser ablation,[22] and machining operations.[23] A wide variety of low-cost polymer materials enable selection for thermal and chemical resistance, molding temperature, and surface derivatization properties.

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In this report, we describe two methods for imprinting microchannels in plastic substrates. We have chosen poly(methyl methacrylate) (PMMA) as the substrate for our devices since it has been reported to be the least hydrophobic of the more common plastic materials available.[24] The first fabrication method utilizes small-diameter wire to imprint channels in plastic substrates to produce microfluid systems with simple linear channel designs. The second method involves imprinting the channels using a design fabricated on a silicon master template, permitting more complicated microfluid channel arrangements. Microchannel devices were characterized by two-dimensional interferometry to assess the reproducibility of the fabrication protocol.

The devices were tested as prototype microfluid devices using electrophoretic pumping for fluid control. To date, there have been very few reports on the electrophoretic properties of plastics.[24-26] Fabrication of simple prototype microchannel devices in plastics will facilitate the investigation of electrophoretic and chromatographic properties of various substrates. Understanding the chemical and electrophoretic behavior in these plastics will provide the basis for the use of these devices in analytical applications.

EXPERIMENTAL SECTION

Reagents. A 50 mmol L^{-1} phosphate buffer with a pH of 7.4 was used in all experiments. 5-(and 6)-Carboxyfluorescein was obtained from Molecular Probes (Eugene, OR), and a 10 mmol L^{-1} solution was prepared in buffer. Polystyrene latex microspheres derivatized with fluorescein isothiocyanate were 0.930 μm in diameter and were obtained from Polysciences, Inc. (Washington, PA). A saturated solution of riboflavin (Sigma Chemical Co., St. Louis, MO) was prepared in distilled water and was used undiluted in the system. Ethylenediamine pyrocatechol (EDP) etchant was obtained from Transene Co., Inc. (Rowley, MA). Human anti-goat IgG conjugated with fluorescein and goat anti-horse IgG were obtained from Pierce (Rockford, IL), and stock solutions were prepared according the supplier's instructions. Final concentrations of human anti-goat IgG and goat anti-horse IgG were 1.3×10^{-5} and 1.3×10^{-4} mol L^{-1}, respectively.

Channel Fabrication with Wire Imprinting. Channels were fabricated using PMMA Plexiglas pieces (blanks) approximately 2 cm in width × 10 cm in length and 1.6 mm thick. Chromel wires (Omega Engineering Inc., Stanford, CT) with diameters of 13 or 25 μm were used to imprint the channels in the plastic substrates. A piece of wire approximately 20 cm in length was stretched taut by clamping the ends to a wire tension bow. The taut wire was placed over the PMMA across the entire blank, and dust was removed from all surfaces using an antistatic brush. The blank and the wire were then placed between two clean microscope slides. The microscope slides holding the PMMA were clamped together between two aluminum blocks that uniformly covered the slides. Then the tension bow was removed by cutting the wire, and the assembly was placed into the oven. The PMMA channels were formed by heating the plastic at 105 °C for 10 min.

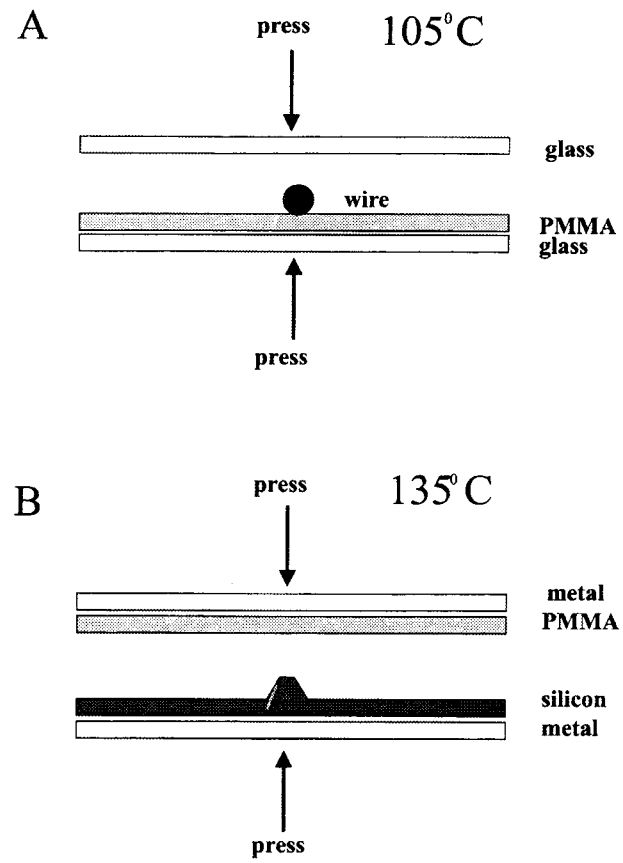

Figure 1. (A) Fabrication protocol for wire-imprinted devices. (B) Fabrication protocol for silicon-template-imprinted devices.

This temperature was chosen on the basis of the reported softening temperature (105 °C) for the polymer. At lower temperatures (90 °C), the resulting imprinted channels were very shallow, and at higher temperatures (120 °C), bubbles were formed in the polymer during the imprinting process. The imprinting procedure is outlined in Figure 1A. The assembly was subsequently removed from the oven and allowed to cool completely. The microscope slides were removed, and the wire was pulled away from the plastic to reveal the channel.

A second channel was made perpendicular to the original channel for the purpose of sample injection. To create this channel, a wire was placed across the width of a second plastic blank of similar dimensions, and the channel was fabricated as described above.

The resulting pieces of plastic, one with a channel across its length and one with a channel across its width, were clamped between two microscope slides and placed in the oven for 10 min. The PMMA devices were heated at 108 °C for 10 min to bond the two pieces together. Prior to bonding, four holes (3 mm in diameter) were drilled through the cover slides. These holes were aligned with the ends of the channels in the imprinted slide to create the buffer, analyte, and two waste reservoirs (wells). A schematic of the completed device is shown in Figure 2.

Design and Fabrication of the Silicon Micromachined Template for Imprinting Channels. The layout of the microfluid channels was drawn using a standard CAD software package. The designs were printed onto a transparency with 50 dots/mm (1270 dpi) resolution, and the transparency image (black on clear) was used as a mask for photolithography.

A 100 nm oxide layer was thermally grown on the surface of a 3 in. (7.62 cm) silicon wafer with crystal orientation ⟨100⟩. A

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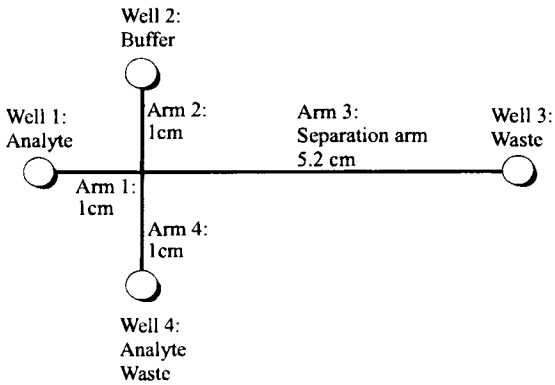

Figure 2. Schematic of prototype wire-imprinted device.

layer of photoresist was spin-coated over the surface of the oxide and subsequently exposed to UV light through the transparency image aligned to the wafer flat. The photoresist was then developed, revealing the transferred image. In this step, all photoresist was removed except for that in the area defining the channels. The wafer was then placed in buffered HF solution to etch the exposed oxide layer. After a thorough rinse with distilled water, the photoresist was removed with acetone. The remaining oxide layer was used as a mask for anisotropic etching of the silicon wafer.

The silicon wafer was etched in EDP for 100 min at 98 °C. Silicon was etched in all areas surrounding the channels, and the resulting structure was a raised three-dimensional image of the channels. This embossed structure was then used to imprint the device image into the plastic substrates.

Preparation of Channels by Imprinting with Micromachined Silicon Template. Channels were imprinted by placing the silicon wafer with the raised image over the plastic blank, the whole assembly being placed between two polished aluminum plates. All surfaces were cleaned with an antistatic brush prior to channel formation. Preparation of the devices required the use of a press to ensure the application of reproducible and uniform pressures to the surface of the silicon wafer. The imprinting procedure is shown in Figure 1B. The two sides of the press could be accurately temperature controlled, and the plastics were heated to 135 °C for 5 min during channel formation. A blank PMMA piece was bonded to the imprinted piece to seal the channels as described for the wire-imprinted devices.

Characterization of Unsealed Microfluid Channels. The microchannel devices were examined by two-dimensional interferometry (Optical Profiler, WYCO Corp., Tuscon, AZ) to determine the dimensions of the resulting channels. The dimensions of devices fabricated by wire imprinting and silicon mold imprinting methods in PMMA were characterized by this technique. These data were used to assess the reproducibility of channel imprinting as well as the quality of channels produced by the different methods.

Characterization of Sealed Microfluid Channels. The maximum width of the sealed microfluid channels was determined by obtaining an image of the device with an imaging system, calibrating the screen image, and measuring the width. To observe the sealing process, microfluid channels were filled with rhodamine-6G-doped UV-curable epoxy. Once the epoxy was hardened, the microchannel was cut using a microtome, and photographs were again obtained using the imaging system.

Hardware for Microchannel Evaluation. A fluorescence microscope equipped with a high-pressure mercury lamp was used to observe the movement of fluorescent reagents in the channels. For detection of fluorescein, emission was measured through a high-pass filter (>515 nm) with excitation through a 390−490 nm band-pass filter. The device was confined on the stage of the microscope by a PMMA holder with four wire contacts, each leading to a separate high-voltage power supply (Model 310KB, Power Designs, Westbury, NY). Each of the four wires was placed into a well containing analyte (well 1) or buffer (wells 2−4). Voltages were applied to each of the arms of the device using four separate power supplies linked to a common ground. The base of the plastic holder was connected to the circuit ground, and the PMMA microfluid device was placed on a piece of grounded aluminum foil to reduce electrostatic charges acquired by the plastic surfaces. Without appropriate grounding, we were unable to control the flow of fluids by choice of the voltages applied to the arms of the device.

Injections of carboxyfluorescein solution were performed by manual manipulation of the applied voltages. Briefly, injections were performed in three steps by applying the following voltages on arms 1−4 of the device. During all three steps, arms 1 and 2 were held at −300 and 200 V, respectively. In step 1, arm 3 was held at ground, and arm 4 was −300 V. In step 2, arms 3 and 4 were both held at ground. In step 3, arm 2 was 450 V, and arm 4 was −300 V. Step 1 was 60 s, step 2 was 10 s, and step 3 was 132 s in duration.

Injections were observed using a CCD camera attached to a videotape recorder and a visual imaging system with a frame grabber board and software capable of capturing 60 images/min (Data Translation, Inc., Marlboro, MA). For quantitation of injected volumes of carboxyfluorescein solution, the CCD camera was replaced with a photomultiplier tube (PMT, Hamamatsu, Bridgewater, NJ). Electropherograms were then obtained by coupling the output of the PMT to a chromatography data system and software package.

Immunoassay Method. Fifty microliters of the stock solution of goat IgG was added to 100 μL of the stock solution of fluorescein-conjugated human anti-goat IgG. The sample solution was placed into the sample well following incubation at room temperature for 15 min. Injections were performed in three steps by applying the following voltages on arms 1−4 of the device. During all three steps, arms 1 and 2 were held at 200 and −200 V, respectively. In step 1, arm 3 was ground, and arm 4 was 300 V. In step 2, arms 3 and 4 were held at ground. In step 3, arm 2 was −1700 V, and arm 4 was 800 V.

RESULTS AND DISCUSSION

Wire-Imprinted Microfluid Channels. Devices were fabricated using the wire imprinting technique with perpendicular channels for sample introduction and analytical separation. Initially, the two channels were imprinted sequentially on the surface of a single plastic blank by first creating the separation channel and, in a separate step, creating the perpendicular channel at a slightly higher temperature (108 °C). However, it was difficult to reproducibly fabricate the cross channel, since the plastic was not easily deformed and seemed to be more rigid following the first heating cycle. Because of this constraint, channels were fabricated in two different plastic blanks that were subsequently sealed together so that the formed structure had an "upper" channel for sample loading and a "lower" channel for separation.

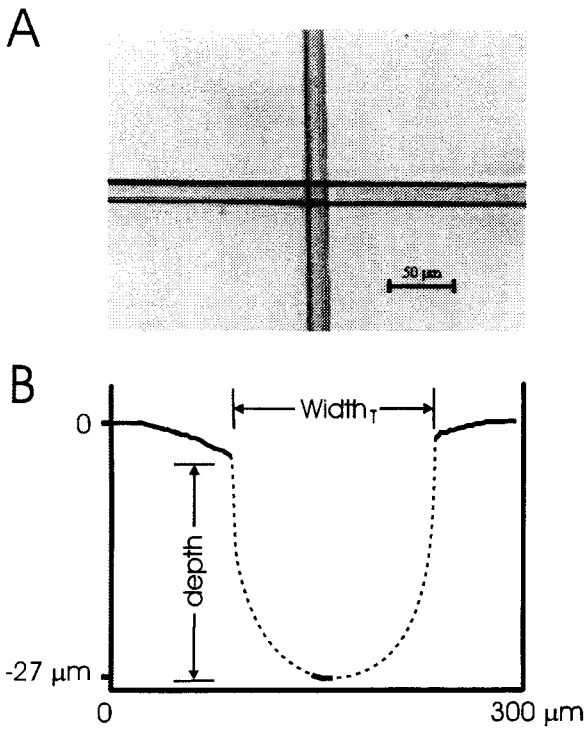

Figure 3. (A) Photograph of a typical 25 μm wire-imprinted device. (B) Typical profile of a wire-imprinted device derived from interferometry and microscopy.

This protocol also helped to reduce variability in channel dimensions on a single device with multiple pressings caused by small differences in temperature, heating time, and clamp pressure. The two channels were produced simultaneously under identical conditions in an attempt to control all factors. The wire-imprinted device is very similar in design to the first-generation microfluidic devices produced in silica-based substrates.[27,28]

To complete the fabrication of the microfluid device, the two pieces of plastic were bonded together to form the sealed channel. This step was accomplished with a high success rate at a slightly higher temperature than the channel imprinting. Although usually successful on the first attempt, some devices were not well-sealed due to bubble entrapment. These failed devices could generally be salvaged by reheating the device for a longer time at the same temperature. A photograph of a typical wire-imprinted device prepared using 25 μm wire is shown in Figure 3A.

Channels were fabricated in PMMA substrates by the wire imprinting technique using two different size wires. Characterization of the formed channels was performed primarily by using two-dimensional interferometry. Two-dimensional interferometry provides detailed information on the surface of the substrate and on the base of the channels. This method provides little or no information on the channel walls; therefore, other procedures must be implemented to observe the wall profile. An optical image of a wire-imprinted device demonstrated that the wire-imprinted channels had the anticipated rounded shape.

The reproducibility of the channel fabrication method was evaluated using the interferometric data. A typical channel profile is shown in Figure 3B for a wire-imprinted device prepared in PMMA. The solid lines in the figure represent the interferometric data used to evaluate the devices. The dotted lines indicate the extrapolated profile observed with microscopy. The channels were evaluated on the basis of the depth of the channels, as well as the width of the channels at the surface of the plastic. These parameters were compared independently, and the results are shown in Table 1. Seven channels were prepared using the 13 μm wire. It can be seen from these data that the depth of the channels is very well-controlled by the wire diameter (rsd = 2.2% for the smallest wire); however, there was considerably more variation in the width of the channel. This relative variability was approximately 42% for the channels prepared using the smallest wire. The average width at the top of the unsealed channel was 3.2 times the width of the wire. It should be noted that, in this procedure, the pressure applied to the device during heating and channel formation was not precisely controlled.

Table 1[a]

substrate, imprinting tool	N	depth (μm), SD	width $_L$(μm), SD	width $_T$(μm), SD
PMMA, 13 μm wire	7	13.4, 0.3		41.7, 17.5
PMMA, 25 μm wire	7	26.7, 0.1		168, 72
PMMA, silicon template	5	43.3, 0.3	32.8, 1.9	421.6, 105.1 378.8, 49.9[b]

[a] N is the number of devices, and SD is the standard deviation of the measurement. Width $_L$ is the width of the channel at the lowest point; width $_T$ is the width of the channel at the top of the channel point. [b] The top profile of one channel was significantly deformed compared with all others. The average width was recalculated without these data.

The channel profile results for seven devices prepared with the 25 μm wire are also shown in Table 1. Again it can be seen that the depth correlates quite well with the wire diameter, with a relative standard deviation among seven channels of 0.4%. The width of the channels did not correlate as well with the wire dimensions. The average width at the top of the unsealed channel was 6.7 times the width of the wire. The relative variability of the measurement of the width at the top of the channel was approximately 43% for the channels prepared using the 25 μm wire.

The variability in the channel width may be explained by the fact that the surface of the plastic was significantly deformed in the vicinity of the imprinted channel. The width and the downward slope of the deformation displayed a significant device-to-device variation. This is depicted in a characteristic profile of an unsealed imprinted channel in Figure 4A. It was found that the temperature and time for sealing the channel were critical. If the sealing time was too short or the temperature was too low, the deformed region in the vicinity of the channel would not seal correctly, as shown in Figure 4B. This microchannel profile can be described to have side "wings" that explain the variation of the data. If the channels were sealed properly, however, the wings disappeared. The well-sealed channel followed more closely the original profile of the wire, as shown in Figure 4C. The average maximum width of eight sealed channels prepared with the 25 μm wire was 32 μm, with a relative standard deviation of 4%. Therefore, most of the variation in the channel profile can be reduced by ensuring that the devices are well-sealed.

The devices fabricated by the wire imprinting technique were simple to produce with minimal equipment. A disadvantage associated with this fabrication method is that the design config-

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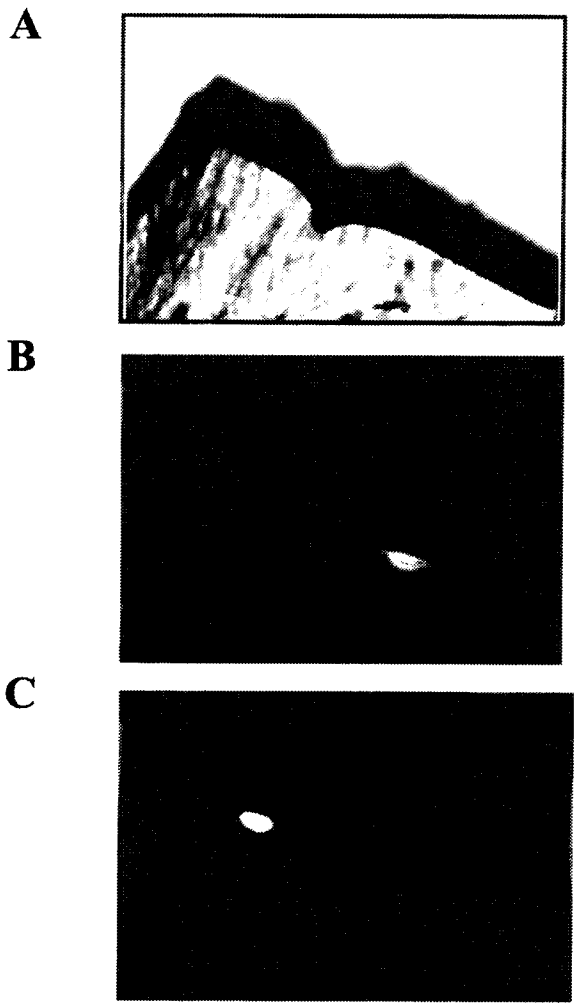

Figure 4. (A) Photograph of unsealed PMMA 25 μm wire-imprinted channel. (B) Photograph of dye-filled wire-imprinted device after sealing for 5 min at 108 °C. (C) Photograph of dye-filled wire-imprinted device after sealing for 10 min at 108 °C.

uration of the fluid channels is constrained in that only simple linear channels may be produced easily with wires. Despite this limitation, useful devices can be made with this technique for the separation of analytes by electrophoresis.

Production of Channels by Silicon Template Imprinting. We have devised a second approach to fabricating imprinted plastic devices for more complicated system designs. Three-dimensional templates were micromachined in silicon and were then used to press channels into plastic substrates. A scanning electron microscopy view of the cross section of the silicon template is shown in Figure 5A, and the interferometric data defining the same device are shown in Figure 5B. The trapezoidal profile of the template is determined by the selective etching of the silicon along the crystal plane. Orientation of the masks along the silicon crystal plane during photolithography was essential for creating uniform silicon templates with small dimensional features. Therefore, the mask was prepared with patterns that were optimized for proper alignment on the silicon wafer during photolithography.

Channels were fabricated by stamping the template to the plastic blank in a press and heating. The silicon template did not adhere to the plastic blank during fabrication and was easily removed following the imprinting procedure. The imprinted devices were quickly removed from the silicon template while still

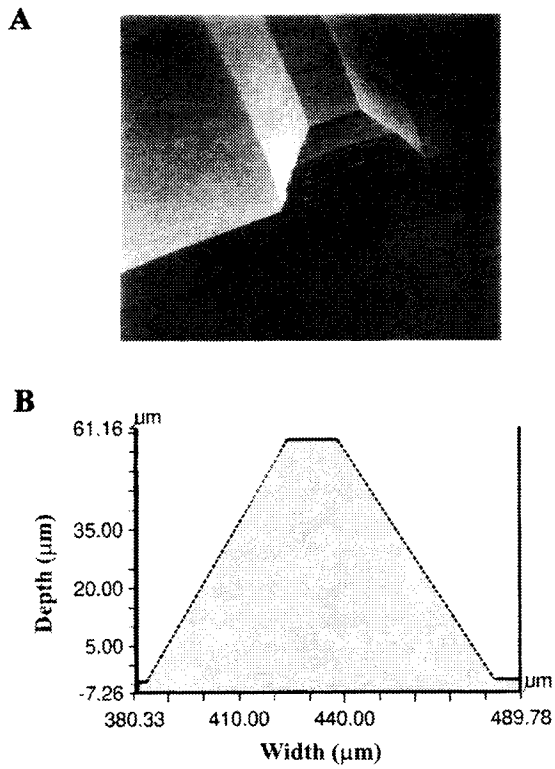

Figure 5. (A) SEM of silicon-micromachined template. (B) Silicon template profile as determined by two-dimensional interferometry. The data on the sides of the template (connecting the top and bottom profiles) are extrapolated from the SEM image.

warm from the press. If the device was allowed to cool against the silicon wafer, the resultant channels were jagged with a stairstep-like appearance that was likely a result of differences in thermal expansion properties of the two materials. If the device was immediately removed from the template during cooling, the resultant channels appeared smooth. The mask used to pattern the template was designed so that two of the arms were offset to create a well-defined sample loop in the device. This is clearly visible in the photograph of the PMMA device prepared by silicon imprinting (Figure 6A).

Effective transfer of silicon templates with small dimensions required the application of uniform pressures across the surfaces during the heating process. The use of a press to produce these devices (instead of the simple clamping device used for wire imprinting) not only improved the channel profile but also reduced the chance of fracturing the template during imprinting due to nonuniform pressure distributions. The interferometric profile and dimensions of a silicon template-imprinted channel are shown in Figure 6B. The highest point in the silicon template used to fabricate this channel (defined by the mask) had the smallest width of 15.5 μm. The silicon was etched to produce a structure 62.3 μm high, and the angle of silicon structure was defined by the anisotropic etching process. The width at the lowest point in the template nearest to the bulk silicon wafer was 92.7 μm. If the structure is tall, the width of the template closest to the bulk silicon will be wide when created by this etching process. A shorter etching time may be used to significantly reduce template dimensions, including the depth and widest width. The narrowest width at the top of the template is defined by the dimensions and quality of the mask. When the image was transferred to the plastic blank, the resultant channel profile had the following dimensions: depth, 61.5 μm; width at the deepest point in the channel,

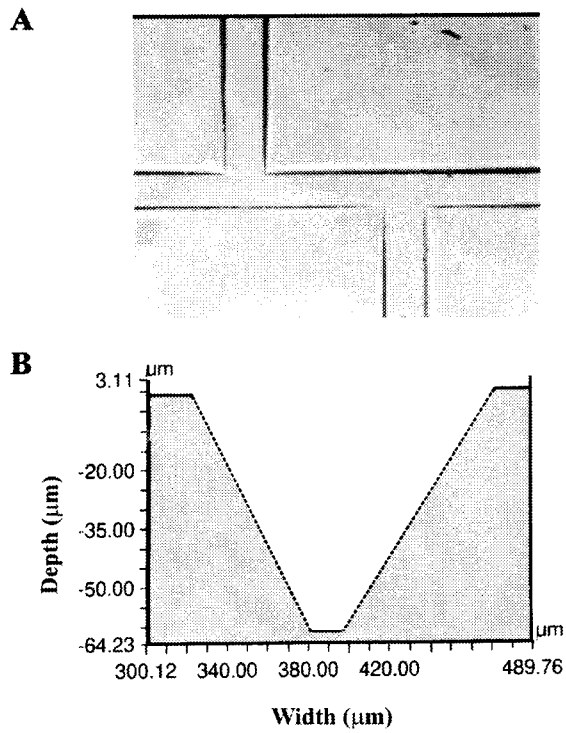

Figure 6. (A) Photograph of silicon-micromachined device. (B) Channel profile obtained by interferometry of silicon-embossed device prepared in PMMA.

Table 2[a]

device	width$_L$ (μm)	width$_T$ (μm)	depth (μm)
1A	43	328	43.5
1B	32	410	43.5
2A	36	258	43.7
2B	31	311	43.0

[a] A delineates the point in the channel farthest from the sample loop at the end of the channel; B delineates the point in the channel close to the sample loop.

15.6 μm; and width at the top of the channel, 169 μm. For a single device, the deviation of the channel profile from the template in terms of depth was 1.3%, for minimum width was 0.7%, and for maximum width was 82.3%.

A second template was prepared with a smaller height to reduce the dimensions of the printed channels. Five devices were prepared using this template, and the resulting profiles were compared to determine the reproducibility of the silicon mold imprinting method (Table 1). The depth and lowest width of the imprinted channels were very reproducible, with low relative standard deviations of 2% and 6%, respectively. The width at the top of the channel had a relative standard deviation of 13%, which is a great improvement over the wire imprinting procedure for unscaled devices.

Two devices were analyzed at two different places along the channel to assess the channel variation on a single device. In both instances, the channels were measured close to the sample loop and then at a point close to the end of the channel. These results are shown in Table 2. Although only two points are reported for each device, it can easily be seen that the within-device channel variation is primarily attributed to differences in the width at the top of the channel.

Testing and Device Applications. Solutions containing fluorescein-coated polystyrene particles or riboflavin were used in the system as "neutral" markers to monitor the direction of the electroosmotic (EO) flow. Although the dye on the surface of the particles should be negatively charged at the pH of the buffer solution, it was assumed that the primary force acting on the particles was the bulk fluid flow, which is dictated by the EO flow. This was supported by the fact that the particles always moved similarly to riboflavin in the device under the same applied electric field. The particles may, therefore, be used to provide qualitative information on the direction of the EO flow but were not considered to be an indicator of the magnitude of the EO flow due to the effect of the charged surface. The EO flow was determined by the injection of the two neutral markers to be from positive to more negative potentials, as was found previously.[24]

The 25 μm wire-imprinted channels were used to demonstrate electrophoretic injections of carboxyfluorescein in the plastic devices. The electrophoretic movement of the negatively charged analyte, carboxyfluorescein, was from negative to more positive potentials. For the injection of carboxyfluorescein, the analyte was placed into well 1 of the device. Fluorescent particles were placed in all wells (1−4) to monitor the direction of the EO flow during the injection of analyte solution. The advantage of using fluorescently labeled particles during the injection protocol is that the particles are a very sensitive indicator of the fluid forces that oppose the electrophoretic movement of the injected analyte. Since materials made from the same polymer may have different compositions that can affect the strength and/or the direction of the EO flow, it is important to rapidly determine the EO flow patterns in the devices in order to perform injections with minimal sample dilution.

The injection protocol was performed in three steps. In step 1, carboxyfluorescein was pumped from well 1 to the center of the cross, as shown in Figure 6A. The EO flow in the device in the first step was strongest from arm 2 to arms 3 and 4, as was determined by the movement and flow of particles around the carboxyfluorescein front. This flow opposed the movement of carboxyfluorescein into other arms of the device. The EO flow from arm 2 to arm 1 was still apparent but appeared to be much lower, since many fewer particles were propelled in this direction. The particles may be seen in Figure 7A as bright spots in the background. In step 2 (Figure 7B), the carboxyfluorescein moved into arms 2 and 4, presumably because of the reduction of (EO) flow across the device as well as the increased electrophoretic flow of the negatively charged dye. In the last step (Figure 7C), an injection of carboxyfluorescein was propelled into arm 3 for analytical separation and evaluation. An advantage associated with this injection protocol is that the size of the sample plug could be altered by varying the time of the second step. This injection protocol, therefore, provides great flexibility with regard to sample size.

There was no evidence of adsorption of the analyte to the surface of the PMMA devices since the channel background fluorescence remained very low and stable throughout a series of sequential injections. The relative standard deviation for peak area for nine sequential injections of carboxyfluorescein was 5.6%. The average retention time for the same peaks was 1.66 min, with an rsd of 1.8%. We did see evidence of adsorption of the particles to the device over time, with notable accumulation of the particles in the center of the injection cross.

The PMMA device was used to demonstrate a prototype immunoassay using fluorescently labeled human anti-goat antibody and unlabeled goat IgG as the antigen. The labeled antibody

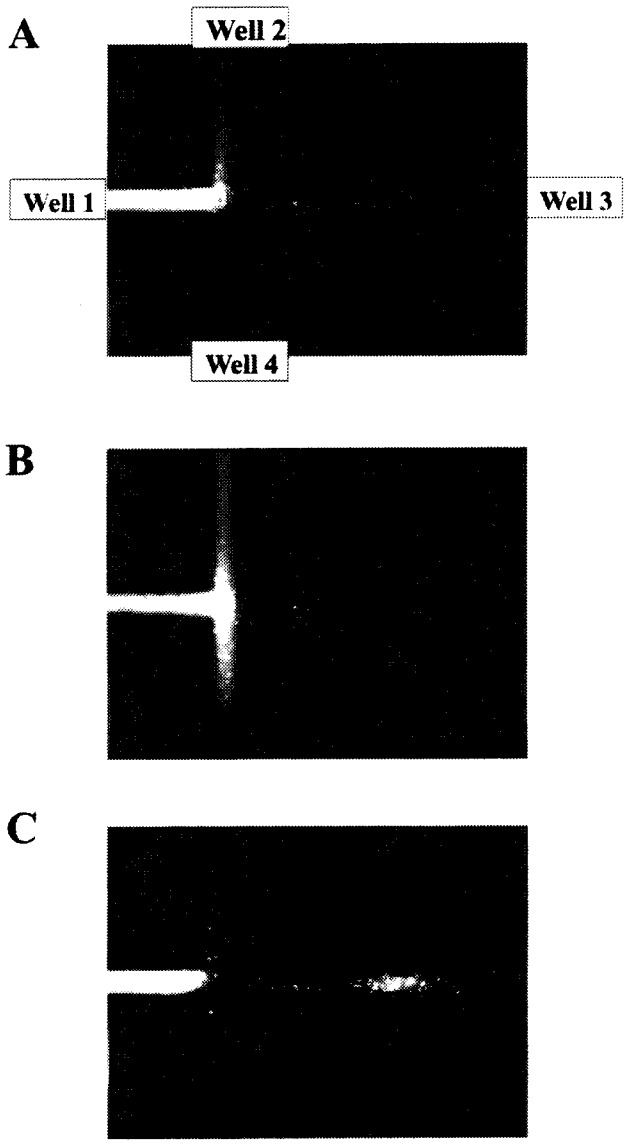

Figure 7. Injection of carboxyfluorescein in wire-imprinted device (description of protocol in text).

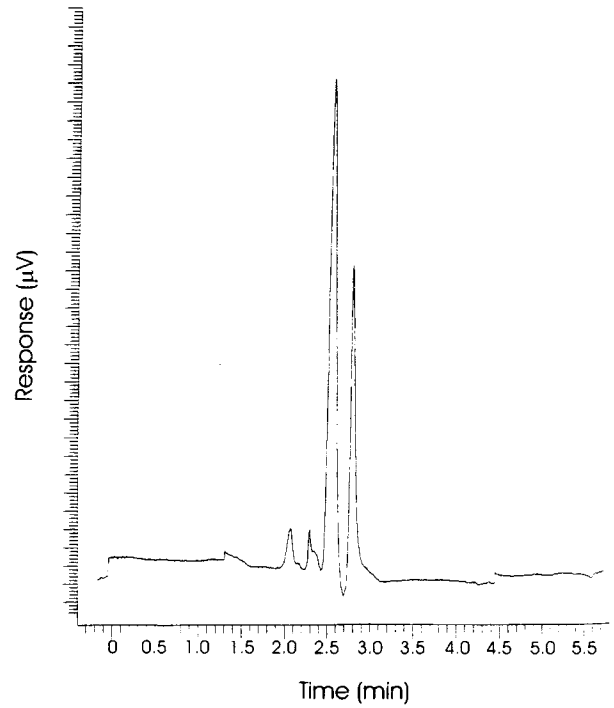

Figure 8. Electropherogram showing results of the immunoassay with human anti-goat antibody and goat IgG as the corresponding antigen. Electrophoretic injection of immunoassay reagents resulted in baseline separation of the assay components in 3 min.

and unlabeled antigen were mixed, incubated, and then injected into the microfluid system by electrokinetic injection. In the microchannel electrophoresis device, the unbound labeled antibody and labeled antibody bound to antigen are detectable. The resulting electropherogram is shown in Figure 8. Baseline separation of the immunoassay components was achieved in 3 min with this protocol. The baseline remained very stable during injections of labeled antibody. There was no indication of protein absorption to the surface of the PMMA device, which may be visualized as baseline drift. The sensitivity and detection limits of this approach are currently under investigation.

CONCLUSIONS

Both wire and silicon template imprinting can be used to produce functional microchannel devices in plastic substrates. The use of a silicon template for the fabrication of plastic microchannels allows for the design of more complex fluid paths compared to the wire imprinting method. In the current fabrication process, the size of the channels is limited by the angle of the anisotropic etch. If higher aspect ratios are required, other silicon etching procedures may be utilized to overcome this disadvantage.

Currently, the field of microfluidics is a high-investment technology requiring the commitment of a fabrication facility for the production of reliable devices. By introducing simple methods for fabrication of microfluid devices in plastic substrates, the possibility exists for rapid growth in basic research in the area of microfluids from both the academic and industrial communities. The technique of wire imprinting should open up opportunities for the inexpensive advancement of this technology. Imprinting more complex fluid circuits using a silicon template involves an initial investment, since the master is micromachined in silicon. However, the overall result is cost-effective production of microfluid devices with potentially complex microfluid designs. The use of micromachined templates should permit rapid production of the next generation of microfluid devices.

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