Use of a micro- to nanochannel for the characterization of surface-enhanced Raman spectroscopy signals from unique functionalized nanoparticles

Brian M. Walton
Po-Jung Huang
Jun Kameoka
Gerard L. Cote
Use of a micro- to nanochannel for the characterization of surface-enhanced Raman spectroscopy signals from unique functionalized nanoparticles

Brian M. Walton,a,* Po-Jung Huang,b Jun Kameoka,c and Gerard L. Cotea,d

aTexas A&M University, Department of Biomedical Engineering, 101 Bizzell Street, College Station, Texas 77843, United States
bTexas A&M University, Department of Material Science and Engineering, 575 Ross Street, College Station, Texas 77843, United States
cTexas A&M University, Department of Electrical and Computer Engineering, 188 Bizzell Street, College Station, Texas 77843, United States
dTexas A&M University, Texas A&M Engineering Experiment Station Center for Remote Health Technologies and Systems, Department of Biomedical Engineering, 101 Bizzell Street, College Station, Texas 77843, United States

Abstract. A micro- to nanochannel nanoparticle aggregating device that does not require any input energy to organize the particles to a specific location, i.e., no pumps, plugs, heat, or magnets, has been designed and used to characterize the surface-enhanced Raman spectroscopy (SERS) signal from four unique functionalized nanoparticles (gold, silver-gold nanocages, silver nanocubes, and silica-gold nanoshells). The SERS signal was assessed in terms of the peak signal strength from the four different Raman reporter functionalized nanoparticles to determine which nanoparticle had better utility in the channel to provide the most robust platform for a future biological analyte detection device. The innovation used to fabricate the micro- to nanochannel device is described; the TEM images of the nanoparticles are shown; the absorption data for the nanoparticles are given; and the spectral data for the Raman reporter, mercaptobenzoic acid (MBA), are depicted. In the micro- to nano-channel described in this work, 5 μl of 22.3 μM MBA functionalized silver nanocubes were determined to have the strongest SERS enhancement.

Keywords: micro- to nano-channel; surface-enhanced Raman spectroscopy; silver nanocubes; gold; silver-gold nanocages; silica-gold nanoshells

Paper 160242PR received Apr. 14, 2016; accepted for publication Aug. 10, 2016; published online Aug. 26, 2016.

1 Introduction

Surface-enhanced Raman spectroscopy (SERS) is an optical technique that can be used to detect analytes at low concentrations.1 Raman spectroscopy provides rotational and vibrational energy information about the molecule adsorbed to the surface, and, because of the significant enhancement factor from the metal surface, SERS can be used for trace analysis.2-7 The enhancements seen from SERS allow examination of molecules in the nano-molar concentration range and below in solution, giving SERS the potential to become a good analytical method for biosensing. The nanoparticles used for SERS can be optimized to help produce a stronger enhancement. Their composition, size, capping agent, and morphology can all be altered to engineer a desired SERS signal.8-11 For example, nanocubes, nanospheres, nanobars, and nanostars are a few of the nanoparticle morphologies that have been engineered specifically to produce SERS enhancements, each having a unique effect on the Raman scattered light.12-17 Silver and gold colloids have also been analyzed using different sizes, preparation methods, and concentrations of analyte or reporter molecules attached to the nanoparticle.18-22

To increase the Raman scattering event probability further, various mechanical methods have been used to make the nanoparticles aggregate together, which causes strong enhancements to occur.23 One mechanical aggregation method was that of Hwang et al., in which an optoelectrofluidic device was used to create plasmonic aggregates aligned at the laser beam spot and produce strong enhancements. However, their device is a complex system that requires an alternating current voltage device to drive it.24 Chrimes et al.25 used a dielectrophoresis approach to trap nanoparticles by changing the frequency and voltage to force nanoparticles to aggregate at certain locations. While this method does produce many enhancements at the desired locations, this approach requires the user to tune the frequency and voltage depending on the size and composition of the nanoparticle.25 Zhou et al.26 have investigated microchannels with a valve system that, when closed, traps the nanoparticles. However, due to the large size of the microchannel with respect to the nanoparticle size, the position of the aggregates is not always in the same location. Therefore, the user has to move the objective around to find the aggregates manually.26 Wang et al.27 developed micro- to nanochannels to capture nanoparticle clusters forming only at the micro- to nanojunction. They were able to produce ~10^8 SERS enhancements at their micro- to nanochannel junction with detection limits down to 10 pM.

In this paper, not only is the design and manufacture of the micro- to nanochannel described, its utility is also demonstrated by using it to analyze and compare colloidal spherical gold,
gold-silver nanocages, silica-gold nanoshells, and silver nano-
cubes in order to determine which nanoparticle has better post-
functionalization utility for providing the best enhancement.

2 Materials and Methods

2.1 Nanoparticle Characterization

Transmission electron microscope (TEM; JEOL JEM-2010
TEM) images of the 60-nm gold spheres, 60-nm silver-gold
nanocages, 100-nm silver nanocubes, and 135-nm silica-gold
nanoshells (Nanocompsix) were collected before functionaliza-
tion to show the nanoparticles’ size and morphology. A Tecan
ultraviolet visible (UV/VIS) spectrometer was used to measure
the extinction spectra of each nanoparticle solution. A NanoSight particle tracking system (NanoSight, LM10, nano-
particle size microscope) was used to characterize the particle
concentrations and particle size distribution. SERS spectra were
also recorded (Thermo Scientific, DXR Raman microscope) to
show the enhancement capabilities of the different nanoparticles
in the micro- to nanochannels. All other reagents mentioned
hereafter were purchased from Sigma-Aldrich.

2.2 Nanoparticle Synthesis

The 60-nm nanocages were obtained from the Xia Group at
Washington University and were prepared using the galvanic
reduction and an AgNP template method as described by
Chen et al. in Ref. 28. Chloroauric acid (HAuCl4) is added to
a solution of Ag nanoparticles, causing the galvanic reduction to
occur, and the nanoparticle takes on the shape of the Ag tem-
plate. The nanocage templates can be engineered to be various
shapes, allowing the colloid’s local surface plasmon resonance
wavelength to be tuned from 500 to 1200 nm based on the molar
ratio of the HAuCl4 to Ag.

The 100-nm silver nanocubes were obtained from the Wu
group at Texas A&M University and prepared from seeding
growth synthesis. They were synthesized from silver nitrate
with copper (II) chloride as a seeding agent in a polyvinylpyr-
rolidone (PVP) pentanedioi solution, as described in Ref. 29.
Ethanol is used to quench the reaction and wash out any remain-
ing reactants. The cubes in the ethanol solution are then diluted
with a PVP/ultrapure solution and filtered. Lastly, the nano-
cubes are washed with ethanol and concentrated to the present
concentration.

The 60-nm gold nanospheres were purchased from Polysciences Inc., and the silica-gold nanoshells were purchased from Nanocomposix.

2.3 Raman Reporter Preparation and
Functionalization to the Nanoparticles

Five milligrams of MBA powder was mixed with 45 ml of 200
proof ethanol and sonicated for 30 min, yielding a 720-μM
MBA/ethanol solution. Another dilution was performed from
the initial stock solution by adding deionized water, yielding
a final MBA concentration of 22.3 μM.

Each of the four types of nanoparticles was mixed at a 1:1
volumetric ratio with 30 μl of 22.3-μM MBA/ethanol solution
and vortexed for ~20 s. After 1 h, the nanoparticles were
washed by adding 200-proof ethanol to the solution to fill up a
1.5-ml centrifuge tube and centrifuging for 30 min. Following
the centrifugation, the supernatant was removed, and deionized
water was added to fill the 1.5-ml centrifuge tube, which was
then centrifuged for 30 min. The supernatant was removed
again and the nanoparticles were resuspended in 60 μl of deion-
ized water to match the initial volume of the nanoparticle MBA
solution to keep the concentration the same. The washing steps
remove the previous capping agent on the nanoparticles, citrate,
or PVP, depending on the nanoparticle, and replace them with
MBA.

2.4 Micro- to Nanochannel Synthesis

The optofluidic micro- to nanochannel device was fabricated
using photolithography and etching. The fabrication process
is shown in Fig. 1. The substrate is a double-sided polished
fused silica wafer with a 500-μm thickness (Mark Optics Inc., Santa Ana, California). For the fabrication of the micro-
channel, 50 nm of silicon nitride (Si3N4) was deposited onto the
fused silica wafer using plasma-enhanced chemical vapor
deposition. The photoresist is then spin-coated on the Si3N4
to make the 20-μm wide microchannel pattern. We used a plastic
mask to make the channels, and the smallest width for plastic
masks is 20 μm. However, it is possible to use a metal mask, and
the smallest width for these is typically 2 μm. After UV expo-
sure, the Si3N4 at the patterned area is etched completely off by
dry etching, leaving only the photoresist on the wafer. Following
this step, the photoresist wafer is submerged into the buffer of
echant, which etches away the exposed silica in the microchan-
nel pattern down to 1.5 μm. Once the inlet and outlet holes for
the microchannel are drilled, the photoresist is stripped off by
acetone and the Si3N4 layer is removed completely by hot phos-
phoric acid (H3PO4, 160°C), which completes the microchannel
fabrication.

For the fabrication of the nanochannel, the photoresist is
spin-coated directly onto a fused silica wafer, and a 20-μm
wide nanochannel is patterned onto the wafer. Afterward,
carbon tetrafluoride (CF4) reactive ion etching is used to make
a 40-μm deep nanochannel into the wafer. Following the

![Fig. 1 Schematic diagram of the fabrication process for the optofluidic device.](image-url)
removal of the photoresist from the nanochannel wafer using acetone, the wafer with the microchannel and the wafer with the nanochannel are thoroughly cleaned via piranha solution and annealed at 1050°C for 10 h. The annealing process permanently bonds the substrates by thermal fusion bonding.

The completed optofluidic device shown in Fig. 2 uses a micro- to nanochannel junction structure to trap the nanoparticles. The microfluidic channel has a depth of 1.5 μm and a width of 20 μm. The nanofluidic channel has a depth of 40 nm and width of 20 μm. The mechanism for introducing and aggregating the nanoparticles in the micro- to nanochannel is shown in Fig. 2(d). After pipetting the solution into the microchannel inlet, the solution is drawn into the optofluidic device by capillary force. All four nanoparticles have diameters that are larger than the 40 nm micro- to nanojunction; therefore, they form clusters at the micro- to nanojunction, which is the SERS detection area.

2.5 Surface-Enhanced Raman Spectroscopy Collection

Micro- to nanochannel SERS spectra were collected using a Thermo Scientific DXR Raman confocal microscope. The samples were excited using a 780 ± 0.2 nm diode laser with a power of 24 mW through a 10x Olympus microscope objective configured with an 1800 grooves/mm grating. Samples were exposed 30 times for 5 s for a total integration of 150 s.

3 Results and Discussion

3.1 Nanoparticle Imaging

Nanoparticle composition, size, capping agent, and morphology are the major components that affect the SERS signal; therefore, we begin by looking at morphology using a TEM image for each nanoparticle. Figure 3 shows the shape and size of the four nanoparticles used in this study. Gold nanospheres were determined to be 60 nm and spherical in shape. Au–Ag nanocages were determined to be 60 nm but are cube-shaped and porous.

The gold-silver nanoshells have a silica core with a gold shell. are 135 nm, and are spherical. The silver nanocubes are 100 nm and cubed in shape. A NanoSight system (Malvern Instruments, Worcestershire, UK) was used to show the concentration, average particle size, and particle distribution for all nanoparticles to verify the particle size and ensure the nanoparticles being compared had the same concentration of 147 pM.

3.2 Extinction Spectra for Nanoparticles

The extinction spectra of the four nanoparticles were analyzed in this study to assess their resonance around the 785-nm laser used in this study. Even if the nanoparticle extinction spectra are not perfectly in resonance with the 780-nm extinction laser, they will be shifted to the near-infrared region through mechanical aggregation. In the absence of mechanical aggregation, this shift has also been achieved by chemical aggregation using NaCl and can also be accomplished using elements that have a positive charge, which attract the negative surface charge of the citrate on the surface of the gold nanoparticles.

However, the SERS experiments discussed in this paper do not need NaCl due to the forced aggregation by the micro- to nanochannel. In addition, as with some of the particles here, the colloidal nanoparticle systems can be engineered to have their extinction spectra tuned to a particular wavelength. For instance, the nanoshells have a dielectric core and are surrounded by a metal shell such as gold or silver, and their extinction spectra are tuned depending on the size and composition of the core/shell ratio. Nanocages can also have their size, and therefore their optical properties, tuned by controlling the molar ratio between the silver template and HAuCl₄ when making the nanocages. For the silver nanocubes, the extinction spectra are tuned primarily by the edge curvature of the nanocubes and particle size. Lastly, gold colloid extinction spectra that do not resonate with the excitation source were chosen to illustrate how the forced aggregation at the micro- to nanochannel shifts the extinction spectra to the desired wavelength to produce SERS signals. The extinction spectra of each type of colloid was collected before and after the addition of MBA, as shown in Fig. 4. As depicted in the data, MBA at this concentration does not cause a shift in the extinction spectra of the four nanoparticles, which is due to the electrostatic affect from MBA that prevents the nanoparticles from aggregating.
Figure 4 also shows that the nanoshells, nanocages, and nano-
cubes are in resonance with the 780-nm excitation wavelength.
The gold nanospheres are the only nanoparticle not engineered
to be in resonance with the excitation wavelength, but a shift in
the extinction spectra maximum peak relative to the excitation
wavelength results in a shift in the enhancement of the corresponding SERS signal when
looking at a single nanoparticle. When using aggregation, the
effectiveness of the corresponding SERS signal increases, but the
aggregation process is non-linear and varies with the concentration
of nanoparticles. Therefore, we discuss capillary flow in the
fluidic flow called capillary flow in this newly designed
microchannel in the x-direction and y-direction. From the equa-
tions, we get the average velocity [Eq. (1)] and time [Eq. (2)]
for the fluid to arrive at the micro-
to nanojunction. The detailed process is described in the Appendix.

\[ u_{avg} = \frac{h^2}{12 \eta w} \left( \frac{1}{L} \right) \left[ \frac{1}{w} + \frac{1}{h} \right] \]

\[ t = \frac{12 \eta L}{2 \sigma \cos \theta} \left[ \frac{h^2}{w} \left( \frac{1}{w} + \frac{1}{h} \right) \right]^{-1} \]

where \( u_{avg} \) is the average velocity, \( x \) is the position in the
x-direction, \( t \) is the time for the fluid to arrive at the micro-
to nanojunction, \( h \) is the height of the microchannel, \( \eta \) is the
viscosity of the fluid, \( w \) is the width of the microchannel, \( \sigma \) is the surface tension, \( \theta \) is the contact angle, and \( L \) is the total
length from inlet to micro- to nanojunction.

Based on Eqs. (1) and (2), the calculated capillary flow velocity
was determined to be 64.9 cm/s, and the time for the fluid to arrive at the junction was 0.153 s. The physical
properties of the fluid, such as viscosity, contact angle, and surface
tension, all remain constant. To improve on Wang’s design,\(^{37}\)
our microchannels were constructed with shallower channels
that were 1.5 \( \mu \)m in depth and 20 \( \mu \)m in width. (Wang’s design
was 6 \( \mu \)m in depth and 150 \( \mu \)m in width.) The velocity in our
new micro- to nanochannel is two times faster than Wang’s
design, and the travel time is also reduced. Our micro- to
nanochannel is designed using an innovative one-step etching pro-
cess rather than multiple etching steps. Previously, two etching
processes, dry and wet etchings, were required for making nano-
channels and microchannels on a single wafer. In this approach,
the two channels are patterned on two different wafers and, as a
result, a single etching process is used to make nano- and
microchannels on each wafer. Hot wet etching is specifically used
to remove the Si₃N₄ layer. One-step etching is advantageous over
the previous method because multiple etching steps cause dam-
age to the surface. Dry or wet etching would create an uneven
surface because energetic plasma or active radicals damage the
surface randomly. Photoresist protection is not effective to
secure the surface from these active chemicals if multiple etching processes are required. Once the surface is damaged,
it will affect the thermal fusion bonding process, which causes
difficulties when trying to permanently bond two substrates.
Therefore, multiple etching procedures should be avoided for
bonding processes. One-step etching also produces a stronger
bond between the two quartz wafers.\(^{38}\) Lastly, these channels
are designed for one-time use only.

3.4 Micro- to Nanochannel Surface-Enhanced
Raman Spectroscopy Analysis

All four nanoparticles were analyzed in the micro- to nanochan-
nel. A solution of 5 \( \mu \)l of functionalized nanoparticles was pipet-
ted into the microchannel inlet, and SERS scans were taken over
time at the nanochannel junction. Each functionalized nanopar-
ticle was evaluated in three different micro- to nanochannels.
Figures 5(a)–5(c) show the micro- to nanochannel junction at
0 min, 5 min, and 25 min, respectively. The horizontal channel
is the microchannel, and the vertical channel is the nanochannel.
At time 0, no nanoparticles have been inserted into the micro
inlet, only deionized water. Figure 5(b) shows the micro-
to nanochannel junction 5 min after 5 \( \mu \)l of the functionalized
silver nanoparticles were pipetted into the micro inlet. The nanoparticles, silver nanocubes in this example, are clearly seen aggregating at the micro- to nanojunction. Figure 5(c) shows the micro- to nanochannel 25 min after the initial 5-μl solution was inserted into the micro inlet, and the larger size of the aggregates formed at the junction can be visualized compared with the 5-min aggregate size. In Fig. 5(d), a Raman scan was taken to show the baseline signal of the micro- to nanochannel. Figure 5(e) shows the SERS spectra at 5 min, with the MBA peaks easily discernible relative to the background spectrum seen in Fig. 5(d). Figure 5(f) shows the increase in SERS signal that occurs after more nanoparticles have moved to the micro- to nanochannel junction. The increase is due to the forced aggregation at the micro- to nanochannel junction, which creates many “hot spots” that form between the nanoparticles, silver nanocubes in this case, that are all in close proximity to each other.8 Lastly, Figs. 6(a)–6(d) show the SERS spectrum of all four functionalized nanoparticles over time at the micro- to nanochannel junction. The main reason the silver nanocubes have the largest enhancement among the four nanoparticles is

![Fig. 5](https://journals.spiedigitallibrary.org/journals/Journal-of-Biomedical-Optics)

**Fig. 5** Images taken with the Thermo Scientific DXR Raman confocal microscope at the micro- to nanochannel junction at various times with the respective silver nanocube SERS signal for that time directly below.

![Fig. 6](https://journals.spiedigitallibrary.org/journals/Journal-of-Biomedical-Optics)

**Fig. 6** The four functionalized nanoparticles over time. Data shown are an average of three SERS scans for 5, 15, and 25 min for nanocages, gold, nanocubes, and nanoshells (a)–(d), respectively.
likely due to the morphology. The cube shape of the silver nanoparticles provides several corners to form hot spots, each giving a significantly increased SERS signal. To calculate the enhancement factor, the Raman spectra of 100-mM MBA dissolved in ethanol was used to compare with the intensity from 22.3 μM of MBA adsorbed to the surfaces of the various nanoparticles. Specifically, enhancement factors for the nanoparticles were calculated using Eq. (3) to be 10^5, 10^3, 10^5, and 10^4 for silver nanocubes, gold nanospheres, gold-silver nanocages, and silica-gold nanoshells, respectively.37

\[ EF = \frac{I_{\text{sers}}}{I_{\text{norm}}} = \frac{C_{\text{norm}}}{C_{\text{sers}}} \]  

where \( I_{\text{sers}} \) is the SERS intensity, \( I_{\text{norm}} \) is the intensity of MBA at 100-mM concentration, \( C_{\text{norm}} \) is the concentration of 100 mM MBA in moles, and \( C_{\text{sers}} \) is the concentration of 22.3 μM MBA in moles.

4 Conclusion

This data shows that, in the micro- to nanochannel, silver nanocubes have a larger enhancement factor than gold nanospheres, silica-gold nanoshells, or gold-silver nanocages. The larger size of the silver nanoparticles contributed to greater SERS intensities due to the larger nanoparticles having more localized surface plasmons propagating on their surfaces, which are in resonance with the excitation wavelength. The cubed shape of the silver nanocubes also allows more hot spots to form than the spherical nanoshells and gold nanocages.38 The nanocubes have several corners to form hot spots when in the vicinity of another nanoparticle, providing more places where the hot spots can occur to yield strong SERS enhancements relative to the spherical nanoparticles. Although the nanocubes have the same cubed shape, the nanocubes give stronger enhancements than the cube-shaped nanocages due to the plasmon damping that occurs between the gold and silver used to engineer the nanocages.38 Altogether, the size, tuned extinction spectra, and many hot spots formed all contribute to the silver nanocubes having the strongest average SERS intensity compared with the other nanoparticles. Overall, four different nanoparticles were analyzed to determine which produces the strongest SERS signal in a micro- to nanochannel. It has been shown that 5 μl of 22.3-μM MBA functionalized 100-nm silver nanocubes exhibited the largest enhancement when using the micro- to nanochannel. Using the SERS method with nanoparticles functionalized with RRRs and microfluidic channels for aggregation has applications for detection of a number of different biomarkers. Specifically, just a few of the blood biomarker experiments that are underway in our group include: the detection of cardiac troponin for determining if a patient has had a heart attack; the detection of bisphenol A to assess toxicity, and the detection of citrulline as a measure of gut function and metabolism.

Appendix

To describe the capillary flow in rectangular microchannels, we combine the Navier–Stoke’s equations [Eqs. (4) and (5)] with the Young–Laplace equation [Eq. (9)] using substitution. From these equations, we get the average velocity [Eq. (12)] and time [Eq. (13)] the fluid takes to get to the micro- to nanochannel junction. The above process is described in Fig. 7.

\[ \frac{\partial u}{\partial t} + u \frac{\partial u}{\partial x} + v \frac{\partial u}{\partial y} = -\frac{\partial P}{\partial x} + \rho g \frac{\partial}{\partial y} \left( \frac{\partial^2 u}{\partial x^2} + \frac{\partial^2 u}{\partial y^2} \right) \]  

Y-direction

\[ \frac{\partial v}{\partial t} + u \frac{\partial v}{\partial x} + v \frac{\partial v}{\partial y} = -\frac{\partial P}{\partial y} + \rho g \frac{\partial}{\partial x} \left( \frac{\partial^2 v}{\partial x^2} + \frac{\partial^2 v}{\partial y^2} \right) \]  

where \( u \) is the fluid velocity in the x-direction, \( v \) is the fluid velocity in the y-direction, \( P \) is the pressure, \( \rho \) is the density of fluid, \( \eta \) is the viscosity of fluid, \( g \) is the gravity, and \( y \) is the time.

Assumption for simplified Navier–Stokes’ Equation: (1) incompressible fluid, (2) steady state, (3) Laminar flow, and (4) Newtonian fluid.

1. Incompressible fluid: The density of the fluid is constant; therefore, the divergence of flow velocity is zero from an equivalent statement (\( \nabla \cdot u = 0 \)).

2. Steady state: The fluid in the microchannel is constant. Therefore, the flow properties would not change with time; \( \partial u/\partial t \) and \( \partial v/\partial t \) are zero.

3. Gravity effects and pressure distribution in the y-direction can be ignored in the microchannel: \( g_x \) and \( g_y \) are zero.

4. The y-direction velocity (\( v \)) does not have flow distribution in the y-direction due to the height being much smaller than the width. According to the flow continuity equation [Eq. (6)], the \( \partial v/\partial y \), \( \partial u/\partial x \), and \( \partial v/\partial x \) are zero

\[ \frac{\partial u}{\partial x} + \frac{\partial v}{\partial y} = 0 \quad \text{(6)} \]

\[ \frac{\partial^2 u}{\partial y^2} = \frac{1}{\eta} \left( \frac{\partial P}{\partial x} \right) \quad \text{(7)} \]

Therefore, Navier–Stokes’ equation is simplified to Eq. (7). The boundary condition in the y-direction is when \( y = (\pm h)/2 \), which gives \( u = 0 \). The solution to Eq. (7) is displayed in Eq. (8)

\[ u(y) = \frac{1}{2\eta} \left( -\frac{\partial P}{\partial x} \right) \left( \frac{h^2}{4} - y^2 \right) \quad \text{(8)} \]
According to the Young–Laplace equation, the capillary pressure ($\Delta P$) in a rectangular microchannel can be described as

$$\Delta P = \sigma \left( \frac{1}{R_w} + \frac{1}{R_h} \right),$$

where $R_w = \left( \frac{w}{2 \cos \theta} \right)$, $R_h = \left( \frac{h}{2 \cos \theta} \right)$. (9)

The capillary flow rate in the rectangular microchannel can be described as

$$u(y) = \frac{1}{2\eta} \left( \frac{1}{x} \right) \left[ 2\sigma \cos \theta \left( \frac{1}{w} + \frac{1}{h} \right) \right].$$

The average velocity of capillary flow in the microchannel can be defined by the volume flow rate in the microchannel

$$u_{\text{avg}} = \frac{Q}{wh} = \frac{h/2}{wh} \int_{-h/2}^{h/2} \left( -\frac{\partial P}{\partial x} \right) \left( \frac{h y - y^2}{4} \right) dy$$

$$= \frac{h^2}{12\eta w} \left( \frac{1}{x} \right) \left[ 2\sigma \cos \theta \left( \frac{1}{w} + \frac{1}{h} \right) \right].$$

Capillary flow can be defined by the physical meaning of velocity, speed, or the average velocity

$$u_{\text{avg}} \equiv \frac{dx}{dt} = \frac{h^2}{12\eta w} \left( \frac{1}{L} \right) \left[ 2\sigma \cos \theta \left( \frac{1}{w} + \frac{1}{h} \right) \right],$$

$$t = \frac{12\eta L}{2\sigma \cos \theta} \left[ \frac{h^2}{w} \left( \frac{1}{w} + \frac{1}{h} \right) \right]^{-1}.$$

where $u_{\text{avg}}$ is the average velocity, $x$: is the position in $x$-direction, $t$ is the time for the fluid to arrive at the micro- to nanojunction, $h$ is the height of the microchannel, $\eta$ is the viscosity of the fluid, $w$ is the width of the microchannel, $\sigma$ is the surface tension, $\theta$ is the contact angle, and $L$ is the total length from the inlet to the micro- to nanojunction.

Using the equations above with the constants below, we find the average velocity and time for the sample to get to the micro- to nanojunction. We also show the average velocity and time for Wang’s paper further below for comparative purposes:

- viscosity of water ($\eta$, 20°C): 1.002 N · S/m²;
- contact angle ($\theta$): 0 deg;
- surface tension ($\sigma$): 72.62 × 10⁻³ N/m;
- length ($L$): 1.5 × 10⁻³ m;
- width ($W$): 20 × 10⁻⁶ m;
- height ($h$): 1.5 × 10⁻⁶ m;
- $u_{\text{avg}} = 64.9$ cm/s;
- time = 0.153 s.

The dimensions of the micro- to nanochannel mentioned in Wang’s paper:

- viscosity of water ($\eta$, 20°C): 1.002 N · S/m²;
- contact angle ($\theta$): 0 deg;
- surface tension ($\sigma$): 72.62 × 10⁻³ N/m;
- length ($L$): 1.5 × 10⁻³ m;
- width ($W$): 150 × 10⁻⁶ m;
- height ($h$): 6.0 × 10⁻⁶ m;
- $u_{\text{avg}} = 33.4$ cm/s;
- time = 0.298 s.

Acknowledgments

Research reported in this publication was supported by the National Institute of Environmental Health Sciences of the National Institutes of Health under Grant No. F30ES023512. The content is solely the responsibility of the authors and does not necessarily represent the official views of the National Institutes of Health.

References

Walton et al.: Use of a micro- to nanochannel for the characterization...