Exposure to live bacteria and accumulation of dead bacteria during bacterialic processes can cause bacterial infectious diseases, implant failure, and antibacterial surface deterioration. Microcapsules with asymmetrically distributed, funnel-shaped pores, which are capable of capturing, retaining, and killing bacteria are developed, offering a solution to bacterial contamination in liquids. It is found that bacterial isolation inside microcapsules is mainly driven by the bacteria’s own motility and the microcapsules’ geometry. After entry into the microcapsule cavity, the bacteria are stably retained inside. The microcapsules shield surrounding cells from exposure to bacterial toxins, as demonstrated by the coculture of rat embryonic fibroblast cells with microcapsules loaded with live Escherichia coli. The microcapsules can be enhanced with a bactericidal coating covering only the interior cavity. This confines the bacteria-killing process, thereby further increasing biocompatibility. The microcapsules may offer a viable bacteria combatant approach as a potentially advantageous method to eradicate bacterial contamination.

1. Introduction

Bacterial infection can lead to hygienic problems,[1] chronic disease, and even mortality.[2] In particular, it is a major cause of medical device-associated infections, which commonly result in medical risks and complications.[3] Therefore, the development of effective antibacterial strategies to isolate the bacteria and thus eliminate the contamination is becoming an increasingly attractive approach to combat bacteria-induced infections.[4] Earlier generations of antibacterial strategy mainly relied on materials impregnated with antibiotics for gradual release to the surrounding tissues, so-called release-based antibiotic systems.[5] Major drawbacks of this strategy are related to their cumulative cytotoxicity and possible increase in toxicity and acute inflammation.[13] Moreover, the inevitable release of bacterial toxin fragments, such as lipopolysaccharides, to the surrounding tissues, thereby causing toxicity and acute inflammation.[13] Moreover, the efficiency of the antibacterial surfaces is adversely affected by nonspecific adsorption of dead bacteria or their fragments.[14] Therefore, the development of novel antibacterial strategies that minimize unspecific adsorption and shield the surrounding tissues from contact with bacterial toxin fragments is desirable but remain a challenge.

To address these challenges, several 2D planar structures have been designed to trap and eradicate motile bacterium from liquid by leveraging their own motility and geometry design.[15] Galajda et al. designed millimeter-scaled open wells connected with funnel-shaped walls to investigate the geometric effects on concentrating motile bacteria. The main outcome of this study was that motile bacteria build up their concentration on the narrow opening side of the funnel wall. However, this technology can only form the structures on the planar 2D surface, in which the microwells are open to the surrounding environment and isolation of bacteria after the concentrating process is difficult. To overcome this problem, Di Giacomo et al. used 3D printing technique to print arrays of 3D microcompartments with funnel-shape channel on planar surface. After printing, the 3D microcompartments are deployed from the planar surface and used to sequester motile bacteria into the confined environment. Despite showing successful sequestering of bacteria inside the microcompartment,[15] it is technically very difficult to integrate the bacteria-killing components into the cavity of the 3D printed trapping systems, since the manufacturing process is not compatible with the loading process of the bacteria-killing components.

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To capture bacteria and combat bacterial lysis out in the open, we here report on a multifunctional and upgradable strategy for eradicating bacteria based on specially designed polymer-based microcapsules with an asymmetric porous geometry. We combined high-throughput droplets microfluidic technology and chemical phase separation processes to successfully design porous microcapsules that enable the effective sequestration of motile bacteria from a liquid. This two-step manufacturing process allows very fine control of the geometry factors of the final microcapsules. The primary feature of these porous microcapsules is their capability to capture, trap, and isolate bacteria inside the cavity of the microcapsules, properties that are pivotal for shielding the bacteria-killing process from the surrounding environment. Through the addition of a bactericidal surface inside the microcapsule cavities by mixing quaternary ammonium silane-modified SiO$_2$ microbeads with polymers, droplet composition quickly reaches the binodal boundary, because of the evaporation of DCM and phase separation of FC oil from the polymer solution. The phase-separated droplets exhibit a core–shell morphology (Figure 1b) consisting of a FC oil-rich core (core phase) and a DCM/polymer-rich shell (shell phase). During this process, PEG chains of PLA-b-PEG in the shell phase diffuse to interfaces and expose the PEG moieties toward the aqueous phase, thereby forming the PEGylated exterior surface layer of the microcapsules (Figure 1b). During the continuous evaporation of DCM, water starts to diffuse into the shell phase, leading to the spontaneous formation of water-in-DCM (W/D) droplets within the shell phase. This is because of Pluronic F-127 forming reverse micelles in DCM, which increases the solubility of water in DCM. Along with the formation of W/D droplets, a dense polymer skin-like layer is formed on the exterior surface of the shell phase due to an increased polymer concentration as a result of DCM depletion on the exterior shell surface. This dense polymer layer inhibits the further diffusion of water and DCM both into and out of the shell phase. Consequently, the trapped W/D droplets within the top layer become the only way for water to continuously diffuse into the sublayer of the shell phase. As a result, the W/D droplets within the top layer are prone to merge to form larger sized W/D droplets (Figure 1b). In contrast, phase separation underneath the shell layer occurs slowly, because lower polymer concentration found there supports long-term stability of the W/D droplets. Consequently, the resulting shell phase

2. Results and Discussion

2.1. Fabrication and Characterization of Microcapsules

A two-step method was established to fabricate microcapsules with a shell that features an asymmetric distribution of funnel-shaped pores (Figure 1). For the first step, we used glass capillary microfluidics for the generation of an oil-in-water emulsion, thereby achieving the segmentation of a continuous polymer organic solution into uniform droplets—the microcapsule templates—in a dispersed phase (Figure 1a). Toward this end, poly(DL-lactide-co-glycolide) (PLGA), poly(L-lactide)-b-poly(ethylene glycol) (PLA-b-PEG), Pluronic F-127, and HFE 7500 oil were dissolved in dichloromethane (DCM) solvent, fed into the middle capillary of the microfluidic device and emulsified with an aqueous solution containing polyvinylpyrrolidone (PVP) surfactant. In the second step, the emulsion droplets were collected and diluted with a large volume of distilled water in the coagulation bath to form microcapsules.

Since HFE 7500 fluorocarbon (FC) oil is poorly miscible with polymers, droplet composition quickly reaches the binodal boundary, because of the evaporation of DCM and phase separation of FC oil from the polymer solution. The phase-separated droplets exhibit a core–shell morphology (Figure 1b) consisting of a FC oil-rich core (core phase) and a DCM/polymer-rich shell (shell phase). During this process, PEG chains of PLA-b-PEG in the shell phase diffuse to interfaces and expose the PEG moieties toward the aqueous phase, thereby forming the PEGylated exterior surface layer of the microcapsules (Figure 1b). During the continuous evaporation of DCM, water starts to diffuse into the shell phase, leading to the spontaneous formation of water-in-DCM (W/D) droplets within the shell phase. This is because of Pluronic F-127 forming reverse micelles in DCM, which increases the solubility of water in DCM. Along with the formation of W/D droplets, a dense polymer skin-like layer is formed on the exterior surface of the shell phase due to an increased polymer concentration as a result of DCM depletion on the exterior shell surface. This dense polymer layer inhibits the further diffusion of water and DCM both into and out of the shell phase. Consequently, the trapped W/D droplets within the top layer become the only way for water to continuously diffuse into the sublayer of the shell phase. As a result, the W/D droplets within the top layer are prone to merge to form larger sized W/D droplets (Figure 1b). In contrast, phase separation underneath the shell layer occurs slowly, because lower polymer concentration found there supports long-term stability of the W/D droplets. Consequently, the resulting shell phase

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**Figure 1.** Schematic illustration showing the two-step strategy for fabricating microcapsules with shells featuring asymmetrically distributed pores. a) Schematic representation of the glass capillary microfluidics device used to form stable oil-in-water droplets. The oil phase in the middle capillary consisted of a DCM solution containing PLGA (to form the shell of the microcapsules), PLA-b-PEG (to PEGylate the microcapsule surface), FC oil (to act as a template to form the hollow cavity of the microcapsules), and Pluronic F-127 (to create pores in the microcapsule shell). The aqueous solution in the outer capillary contained PVP surfactant. b) Schematic representation and the bright field images of various steps of the microcapsule formation process, scale bar = 10 μm.
consists of a dense skin-like outer layer connected to a porous supporting sublayer. Finally, freeze-drying removes FC oil from the core of the microcapsules, forming microcapsules with hollow cavity. Their shell features a PEGylated exterior surface supported by an inner shell structure interspersed with asymmetrically distributed, funnel-shaped pores (Figure 1b). To investigate the porous structure of microcapsules by confocal laser scanning microscope, the microcapsule shells were labeled with Nile red. As shown in Figure 2a, the microcapsules were homogeneous in size (53.8 ± 6.1 µm) and shell thickness (5.98 ± 0.9 µm) (Figure S1, Supporting Information). Moreover, it can be observed that the microcapsules’ shell consists of a thin and dense exterior layer which is supported by a porous sublayer (Video S1, Supporting Information). Cryo scanning electron microscopy (cryo-SEM) was employed to investigate the shell pores structures (Figure 2b,c). The micrographs of the freeze-fractured microcapsules show that the exterior surface of the microcapsules is approximately twice less porous than the interior surface facing the hollow cavity (Figure 2b–d; Figure S2, Supporting Information). The pore structure of the microcapsule shell can be expected to play an essential role in bacteria uptake. Therefore, a z-stack of fluorescence images was generated and the pore structure on each slice analyzed (Video S2 and Figures S2 and S3, Supporting Information). The analysis revealed that the microcapsule shell is mainly composed of four kinds of porous structures, two of which cross the entire exterior layer and extend to the interior hollow cavity (hitherto referred to as translayer pores) and two of which do not (see Figure S3 in the Supporting Information for the schematic representation). The incidence of each kind of pore features within the microcapsules shell was quantitatively analyzed and summarized in Table S3 in the Supporting Information. The quantitative analysis revealed that 21.2% are translayer pores that consist of a large entrance cavity on the exterior surface from which one or more narrow channels extend to the interior surface. These funnel-shaped channels are averagely distributed within the whole microcapsule shell, increasing the access points to the motile bacteria. The other type of translayer pores (16.7%) exhibit narrow canal-like features with a uniform diameter. The nontranslayer pores are consisting of pores embedded in the microcapsule shell that have no outside connection (25%) and cul-de-sac pores with only an opening to the interior surface (37%) (Table S3, Supporting Information).

Raman spectroscopy and zeta potential measurements were applied to investigate the chemical configuration of the microcapsule shell. Analysis of Raman spectra (Figure S4, Supporting Information) revealed that the microcapsule shell consists of...
both PLGA and PLA-b-PEG components. Additionally, zeta potential measurements revealed that the negative surface zeta potential of the microcapsules decreased after addition of PLA-b-PEG. Microcapsules consisting of 100% PLGA showed an electrophoretic potential of $\pm 20$ mV. In contrast, microcapsules formed by blending PLGA with different weights of PLA-b-PEG showed a zeta potential value of $\pm 5$ mV (Figure 2f), indicating the successful incorporation of PEG chains into the microcapsules' exterior surface. To assess microcapsule surface properties with regard to their resistance to nonspecific adsorption, we performed a quantitative analysis of bovine serum albumin (BSA) adsorption (Figure 2g). Microcapsules formed by blending PLGA and PLA-b-PEG (10:1) were less prone to BSA adsorption (1.5 ± 0.6 µg cm$^{-2}$) than microcapsules formed using pure PLGA (7.9 ± 0.8 µg cm$^{-2}$) (Figure 2g; Table S1, Supporting Information).

By regulating the polymer precipitation rate, microcapsules with different shell thickness can be formed. These physical properties, in turn, affect mechanical properties of the microcapsules.[21] To assess mechanical properties, indentation measurements were performed using two groups of microcapsules with a similar size but differing in shell thickness: thick-shelled microcapsules (5.98 ± 0.9 µm) and thin-shelled microcapsules (2.3 ± 0.7 µm, Figures S5 and S6, Supporting Information). Load–displacement curves of 75 individual microcapsule each group were collected by pressing each microcapsule between a moving flattened tip ($d = 50$ µm) and the test stage. A similar shape of the load--displacement curves was obtained for both microcapsules (Figure S7, Supporting Information). Yet, the Young’s modulus and the hardness values derived from the load–displacements curves[22] are different. The thin-shelled microcapsules have an average Young’s modulus and hardness values of 70 and 2.96 MPa, respectively. In comparison, thick-shelled microcapsules have higher Young’s modulus of 210 MPa and a hardness value of 7.86 MPa, indicating that a thicker polymeric shell endows microcapsules with a greater ability to resist deformation (Figure S8, Supporting Information). As a second drawback, the thin-shelled microcapsules also had a less porous shell. Therefore, fabrication conditions were optimized to create microcapsules with thick shells featuring a large number of asymmetrically distributed, funnel-shaped pores for further studies.

### 2.2. Entry of Bacteria into Microcapsules

To assess the ability of the microcapsules to capture and isolate bacteria, a green fluorescent protein (GFP)-expressing *Escherichia coli* were mixed with a Nile red-labeled microcapsule suspension and incubated for 10 min under gentle shaking at room temperature prior to investigation (Figure 3a). A 3D reconstruction view of the microcapsules was created to study the distribution of *E. coli* within the bacteria/microcapsule mixtures (Figure 3a; Video S3, Supporting Information). This analysis revealed that free *E. coli* were located mainly along the glass slide surface (rectangles indicated). Higher up in the 3D reconstruction image, *E. coli* can be observed on the exterior surface of the microcapsules in close proximity to pores and while entering pores (circles indicated). Cross-section analyses of microcapsules (Figure 3a) revealed that, in addition to *E. coli* trapped inside the microcapsule cavity, a number of *E. coli* were visibly aligned within porous areas of the microcapsule shell (Figure 3a; Figure S9, Supporting Information). These observations confirm entering of *E. coli* into the microcapsules by way of the pores.

To study the entry of *E. coli* into microcapsules in a real-time manner, the bacteria and microcapsules were successively added to the observation chamber and observed with time-lapse microscopy (Video S4, Supporting Information). Real-time analysis confirmed that the entry of *E. coli* occurred through the translayer pores of the microcapsule shells. It was noted here that *E. coli* did not adhere to the microcapsules’ exterior surface. Additionally, time-lapse analysis revealed that free-swimming *E. coli* facing away from the microcapsule pores during locomotion, simply passed the microcapsule. In contrast, *E. coli* facing...
2.3. Effect of Bacterial Motility on Entry into Microcapsules

It has been reported that motility of *E. coli* is essential for their squeezing through narrow constrictions.[23] Considering that no chemically attracting agents are presented in the microcapsules, entry of *E. coli* seems to be mainly driven by the bacteria’s own motility. To assess the effect of bacterial motility on entry into the microcapsules, we compared the behavior of motile *E. coli* with nonmotile bacteria, *Staphylococcus sciuri*, under similar conditions. We incubated identical amounts of microcapsules with a similar concentration of either *E. coli* or *S. sciuri* for designated time lengths. After each incubation time period, the microcapsules were centrifuged and washed several times to remove any free bacteria. Finally, the number of bacteria inside the microcapsule shells and cavities were counted. The obtained series of time-lapse microscopic images showed that most *E. coli* were trapped inside the hollow cavity of the microcapsules. In contrast, *S. sciuri* were mostly located inside the microcapsule shell pores (Figure S10, Supporting Information). Statistical analysis revealed that the number of bacteria trapped within the microcapsule shell pores was smaller for *E. coli* than for *S. sciuri* (Figure 4a). Moreover, for every incubation time point at least four times more *E. coli* than *S. sciuri* were found inside the microcapsule cavity (Figure 4b). This indicates the importance of bacterial motility for penetrating the microcapsules.

Further time-lapse microscopic observations were carried out to track the motion path of *E. coli* and *S. sciuri* as they traverse the microcapsule shell toward the hollow interior cavity (Figure 4c,d; Video S5, Supporting Information). This analysis revealed two major differences relating to bacterial displacement and the directionality of bacterial motion. *E. coli* trapped in microcapsule shell pores (indicated by arrows in Figure 4c, right image) showed a larger displacement predominantly directed toward the microcapsule's hollow cavity. Once inside the cavity, the *E. coli* exhibited free swimming with motion paths covering most of the interior microcapsule cavity (Figure 4c, right image). In contrast, *S. sciuri* showed negligible displacement on overwhelmingly nondirectional motion paths (Figure 4d, right image). Most *S. sciuri* trapped within the microcapsule shell hovered in their original position. Those able to reach the microcapsule's interior cavity remained in close proximity to the cavity surface (Figure 4d, right image; Figure S10 and Video S5, Supporting Information). The results indicated that the motility may be one of the important factors to differences in how *E. coli* and *S. sciuri* are captured by microcapsules. Unlike passing through smooth pore surface in which *E. coli* move more effectively and is prone to slide along the pore wall in a smoother motion path,[15] *E. coli* may adopt a more complicated motion manner when passing through the pores of the microcapsules as that observed in porous hydrogels and soil system.[24]

Due to the convexity on the curved surface of the microcapsule’s pores, *E. coli* will intermittently and transiently be trapped when passing through the pores. When *E. coli* are trapped, they may constantly reorient the body until they are able to escape; they then move in a directed path through the pore channel space until they again encounter another curve surface in the channels and until they finally enter the cavity of the microcapsules.[24]

Considering the important role of bacterial motility, it would be interesting to systematically study the degree of *E. coli*’s motility on their entry into the developed microcapsules. Genetical manipulation can be a powerful tool to systematically control the *E. coli*’s motility, such as their motion speed and direction.[25] Therefore, genetically modified *E. coli* will be very interesting candidates for further studying the effects of bacterial motility on penetration into the porous materials. Moreover, other biophysical properties such as shape, size and aspect ratio may also affect bacterial motility through the porous structure of microcapsules.[23b] Furthermore, chemical composition of the bacterial cell wall will determine the mechanical properties of bacteria and will ultimately affect the bacterial entry into the microcapsules. For example, it has been shown that the Gram-positive *Bacillus subtilis* possess an inferior ability to enter small pores than Gram-negative *E. coli*, although both types of bacteria have similar physical properties and similar motility.[23a] Therefore, when considering the application of
microcapsules to sequester bacteria, analysis of bacterial cell wall chemical properties should be taken into account.

2.4. Loading Capacity of Microcapsules

To study the effects of bacteria concentration on the loading efficiency of the microcapsules, identical amounts of microcapsules were incubated with different concentrations of *E. coli* over a time period of 10 min. The analysis of confocal microscopy images revealed that the number of *E. coli* trapped inside the microcapsules decreased parallel with the concentration of *E. coli* in the sample, thus highlighting the role that statistical probability plays in the ability of bacteria to find and enter into microcapsule pores (Figure 5a–e). However, it is important to note that, an accumulation of bacteria was observed inside the microcapsules compared to the outside environment (Figure 5a–e). Statistical analysis performed on these microcapsules confirmed these observations (Figure 5f). Moreover, for the bacteria concentration (≥0.59 × 10⁸ bacteria per mL) the concentration of *E. coli* per milliliter inside the microcapsules after ten minutes was significantly higher than the concentration of *E. coli* prior to adding the microcapsules. This observation clearly shows that the microcapsules are capable of concentrating *E. coli* from the medium without the necessity of chemo-attractive agents. A potential application for using these microcapsules to remove bacteria from an aqueous environment was demonstrated by mixing a fixed amount of *E. coli* with different amounts of microcapsules and incubating the mixture for ten minutes (Figure 5g,h). It was found that the addition of increasing amounts of porous microcapsules has a decreasing effect on the concentration of *E. coli* in the medium.

2.5. Retention of Bacteria in Microcapsules

As a next step, we investigated how motile bacteria are retained inside the microcapsules once they have accumulated after 10 min incubation, especially considering the continuous external water and shear flow in the medium. Toward this end, we studied the ability of the microcapsules to retain captured *E. coli* during exposure to repeated centrifugation and washing steps. Microscopic observations revealed that all uncaptured *E. coli* in the medium were washed away and no bacteria were observed on the external microcapsule surfaces (Figure 6a). Importantly, the number of bacteria inside the microcapsules remained relatively constant for over three repeated washing and centrifugation steps, demonstrating the ability of the microcapsules to protect the captured bacteria from external physical forces generated by the centrifugation and washing steps (Figure 6b). These results suggest that the microcapsule shell provides a stable protective barrier that is able to both isolate the encapsulated *E. coli* from external environmental conditions and to prevent bacterial leakage. Furthermore, a microfluidic flow chamber was designed to study the effect of external flow on the ability of the microcapsules to keep *E. coli* inside and to also compare the motion paths of the loaded and the free-moving *E. coli* in real time (Figure 6c,d). The motion tracking analysis of the bacteria motion paths indicated that the encapsulated *E. coli* were not affected by the external flow as they exhibited...
a random and irregular motion path restricted to the interior of the microcapsule cavity (Figure 6d, right image; Video S6, Supporting Information). In contrast, free \textit{E. coli} outside the microcapsules displayed greater displacement with a longer, straighter and smoother motion path following external flow in the flow chamber (Figure 6d; Video S6, Supporting Information). This analysis confirmed that the microcapsule shells are able to shield the encapsulated \textit{E. coli} from external flow conditions and to retain caught \textit{E. coli} inside the microcapsules.

To investigate the retention of bacteria inside the microcapsules over time, the number of encapsulated \textit{E. coli} inside the microcapsules was continuously monitored for 24 h under gentle minimal medium flow (Figure 6e; Video S7, Supporting Information). It was found that the bacteria kept their active motility while taking advantage of the entire microcapsule cavity area to move around. Despite their motility, most bacteria remained within the microcapsules over 24 h of observation (Figure 6f), indicating the ability of the microcapsules to provide long-term \textit{E. coli} retention. This may be due to the small internal diameter of the channels that could reduce the escape of trapped bacteria from the cavity, indicating the superiority of our technique to the 3D printing technique that can only form bacteria trap with large dimension of funnel-shaped channel.\cite{15} Bacterial growth of \textit{E. coli} within the microcapsules (using bacteria without GFP expression) was studied by culturing \textit{E. coli}-loaded microcapsules in full bacterial growth medium. Standard \textit{E. coli} growth inside microcapsules was observed over time and quantified using a LIVE/DEAD BacLight Bacterial Viability Kit, which stains live bacteria with intact membranes green and bacteria with compromised membranes red (Figure S11, Supporting Information).

2.6. Biocompatibility Evaluation of Bacteria-Loaded Microcapsules

The developed microcapsules are mainly composed of PLGA that exhibits a very good long-term chemical stability in the physiological conditions.\cite{26} These properties allow for testing the developed microcapsules for their ability to shield...
the surrounding environment and eukaryotic cells from bacteria and their toxins for further potential biomedical application. Toward this end, eukaryotic rat embryonic fibroblast (REF) cells were cultured with microcapsules containing live *E. coli*. As a control similar REF cells were also cultured with free live *E. coli*. LIVE/DEAD cell viability assays showed that all REF cells cultured with free live *E. coli* were dead after 21 h, since it does not stain positive for the LIVE-dye (Figure 7a). In contrast, 78.8 ± 31.2% of the fibroblast cells cultured with *E. coli*-containing microcapsules remained viable, as indicated by the positive LIVE staining. (Figure 7b; Figure S12, Supporting Information). These results indicate that the microcapsules are capable of reducing the toxic effects of bacteria on cell survival. It should also be noted that following 28 h of culture, the viability of REF cells cultured with microcapsules containing live *E. coli* was reduced to 43.5 ± 16% (Figure S12, Supporting Information). This loss of cell viability might be attributed to minor leakage of live *E. coli* from the microcapsules into the cell culture medium.

To avoid possible leakage of live bacteria from the microcapsules, microcapsules with an integrated bacteria-killing system were developed. Toward this end, quaternary ammonium silane-modified SiO$_2$ microbeads were produced and encapsulated into the microcapsules during the formation process (Figure 7c,d, Figure S13, Supporting Information). OQAS is a hydrophobic cationic compound that is widely used as an antibacterial coating promoting bacterial adhesion and lysis.$^{[27]}$ Prior to encapsulating OQAS-SiO$_2$ beads into the microcapsules, their functionality was tested in an *E. coli* suspension (Figure S14, Supporting Information). Following the incubation of *E. coli*-containing microcapsules loaded with beads (Figure 7c), *E. coli* viability inside the microcapsules was determined using the LIVE/DEAD bacterial viability assay. The results revealed that *E. coli* contact to OQAS-SiO$_2$ microbeads resulted in efficient killing of bacteria (Figure 7d,e). Next, the motility path of *E. coli* trapped in bead-containing microcapsules was tracked. *E. coli* showed only negligible displacement after contact with the OQAS-SiO$_2$ microbeads inside the microcapsules (Figure 7f). In contrast, *E. coli* inside microcapsules without OQAS-SiO$_2$ microbeads roamed all around the microcapsule cavity (Video S8, Supporting Information). These results indicate that the encapsulated OQAS-SiO$_2$ microbeads can adhere to and lyse the coencapsulated bacteria, and, quite importantly, retain the dead bacteria and, most probably, their fragments in the hollow microcapsule cavity. To assess that, we investigated the biocompatibility of the microcapsules.
Supporting Information

Supporting Information is available from the Wiley Online Library or from the author.

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Conflict of Interest

The authors declare no conflict of interest.

Keywords

antibacterial materials, microfluidics, polymers, porous microcapsules
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