Probing cell surface interactions using atomic force microscope cantilevers functionalized for quantum dot-enabled Förster resonance energy transfer

Zhe Sun,a Amee Juriani,b Gerald A. Meininger, and Kenith E. Meissnerb,*

aUniversity of Missouri, Columbia, Dalton Cardiovascular Research Center and Department of Medical Pharmacology and Physiology, 134 Research Park Drive, Columbia, Missouri 65211
bTexas A&M University, Department of Biomedical Engineering, 337 Zachery Engineering Center, 3120 TAMU, College Station, Texas 77843

Abstract. Förster resonance energy transfer (FRET) between quantum dot (QD) donors and red fluorescent protein (RFP)-tagged integrin acceptors in live cells is reported for the first time. A silica microsphere was coated with CdSe/ZnS QDs and mounted to the cantilever of an atomic force microscope (AFM). The QD microsphere is then conjugated with fibronectin to bind with RFP-αv integrins expressed on the surface of HeLa cells. Following AFM-controlled cell contact with the QD-microsphere structure, FRET is observed between the QD-RFP pair using a photobleaching measurement technique. This FRET probe technique provides a novel tool for studying the cell surface receptor-ligand interactions in biomedical and biological research.

Keywords: Quantum dots; real-time imaging; fluorescence; microscopy; cells; scanning microscopy.

Paper 09053LRR received Feb. 19, 2009; revised manuscript received May 27, 2009; accepted for publication May 28, 2009; published online Jul. 10, 2009.

1 Introduction

Integrins are cell adhesion molecules that play a central role in cell interaction with the extracellular-matrix (ECM). Integrin-ECM interactions determine many fundamental processes, such as cell adhesion, spreading, migration, and growth. The initial integrins binding with ECM will form small dot-like clusters, which could then either mature into focal adhesions or disassemble and disappear in minutes, suggesting rapid regulation of integrins by chemical and/or physical signaling. Novel optical techniques are needed to address the temporal and spatial aspect of integrin-ligand interactions in lifetime with improved resolution.

Development in Förster resonance energy transfer (FRET) techniques have facilitated the study of protein-protein interactions in recent years. This energy transfer is the result of long-range dipole-dipole interactions and takes place only when the two molecules are within 20–100 Å. This distance constraint allows FRET techniques to provide extremely high spatial resolution for resolving molecular interactions.

Semiconductor quantum dots (QDs) have been effectively applied as FRET donors both as free particles as well as when attached to a larger structure, such as an atomic force microscope (AFM) tip or a microsphere. QDs have broad excitation spectra that increase to shorter wavelengths and narrow, symmetric photoluminescence emission spectra (25–45 nm) that can be size-tuned through the visible and into the near infrared. These unique properties enable optimization of FRET with any chosen acceptor fluorophore by minimizing both acceptor excitation and overlap between donor and acceptor emission. Finally, QDs possess relatively high quantum yield and high resistance to photobleaching. This feature makes QDs optimal for use as the FRET donor for multiple measurements. Here, we report a QD-based FRET technique that enables the induction of focal contacts/ adhesions and detection of fibronectin (FN)-integrin interactions on the cell membrane of a live cell.

2 Experimental

CdSe QDs were fabricated using methods described by Peng and Peng. Triocetyolphosphine oxide, cadmium oxide, and a stabilizer were heated to 340 °C under argon for 40 min in a two-neck flask, then cooled to 280 °C. A Se-trioctylphosphine stock solution was injected to start QD synthesis. At a time point corresponding to the desired emission wavelength, the reaction was stopped by removal from heat. To make the QDs soluble in water, QDs in chloroform at 2.5 gm/L were mixed in a 1:1 ratio with butanol and 1 mol/L dihydrolipoic acid, and incubated at room temperature for 24 h. To coat silica microspheres with a layer of QDs, 3-APS-functionalyzed silica microspheres (diameter ~5 μm) are mixed with the DHLA-coated QDs in butanol under agitation. Free QDs are removed by gravity sedimentation procedure.

QD-coated microspheres (QD-microspheres) are attached to AFM cantilevers (spring constant 0.01–0.015 N/m, Veeco Metrology, Inc.) using epoxy (Progressive Epoxy Polymers Inc.). A drop of the microsphere-butanol solution and a drop of epoxy are applied side-by-side to a thoroughly cleaned slide, and the butanol evaporated. An AFM cantilever is first dipped into the epoxy and then lowered onto a QD-microsphere. On withdrawal of the AFM probe, the QD-microsphere sticks to the cantilever and the epoxy cures in 24 h. Immediately before the FRET experiment, the QD-microsphere on AFM cantilever is immersed in 10 μL of FN or bovine serum albumin (BSA) solution (0.5 mg/mL) for 8 min, then washed 4 times with phosphate buffered saline.

HeLa cells are cultured in minimal essential medium (MEM) with 10% fetal bovine serum in a humidified incubator with 5% CO₂ at 37 °C. All culture reagents are purchased from Invitrogen (Carlsbad, CA). For FRET experiments, cells are transfected with construct encoding integrin αv-red fluorescent protein (RFP) (Evrogen Inc., Russia) in 50-mm
dishes with a number 1 glass coverslip bottom (World Precision Instruments, Inc., Sarasota, FA). For transfection, HeLa cells are plated at 50–70% confluency overnight. 6 μL Turbofectin reagents (Origene, Inc.) are first mixed with 100 μL MEM at 25°C for 5 min. 2 μg DNA are then added to the mixture, and incubated for 20 min. The mixtures are then added to the cells. The expression of integrin αv-RFP is usually observed within 24–48 h.

FRET measurements were performed with an Olympus Fluoview1000 confocal microscope. The FRET donor, QDs are excited at 440 nm and the emission collected at 520–550 nm. ~53% of the QD emission spectra [Fig. 1(b)]. There was no significant photobleaching of QDs over the data acquisition period. The FRET acceptor, RFP is excited at 561 nm, and the emission collected 580–680 nm [Fig. 1(b)]. There was minimal donor breakthrough into the acceptor channel [Fig. 2(d)].

3 Results

In this assay, the AFM lowers the FN-QD-microsphere onto the cell surface in order to interact with integrin αv-RFP expressed on cell surface. Figure 1(a) is a concept sketch of the technique. Figure 1(b) illustrates donor emission and acceptor absorption spectra used in this experiment. The calculated Förster distance, \( R_{0} \), at the FRET efficiency of 50% is 5.1 nm. The QD layer on the microsphere was easily detectable under fluorescent microscopy [Fig. 1(d)]. As opposed to embedded QDs, the surface layer of QDs minimized the background noise signal of donor emission. As a proof of concept, FRET with an efficiency of 25–40% was previously observed between QD-microsphere structures and 3 integrins immunolabeled with Alexa546 on fixed cells. In the current study, the RFP is tagged on the c-terminal of αv integrins and resides on the inner leaflet of the cell membrane. Thus, the cell membrane separates the donor and acceptor by approximately 3.5–5 nm. However, we demonstrate that the FRET signal is still experimentally observable.

The stability of the QD coating on the microsphere was confirmed by bringing the QD-microsphere into contact with the cell surface followed by retraction using AFM. No QDs were observed on the cell surface after microsphere retraction. For the FRET experiments, the FN-QD-microsphere was controlled by the AFM to stably engage with cell surface (contact force approximately 500–750 pN) for 20 min to induce FN-integrin interactions. We have previously shown that 20 min is sufficient to induce the formation of a focal adhesion site. A photobleaching technique was applied for measurement of the FRET efficiency. FRET signal manifested as an increase in donor emission following photobleaching of the acceptor. Eight initial images of the donor and acceptor were acquired sequentially and averaged [Figs. 2(a) and 2(b)]. The acceptor was then photobleached over a period of ~3 min. Eight final images of the donor and acceptor were acquired and averaged [Figs. 2(c) and 2(d)]. After photobleaching, the RFP no longer shows fluorescence and the absence of a donor breakthrough in Fig. 2(d) is noted. A comparison of Fig. 2(a) 2(c) shows increased luminescence from the QD-microsphere. The FRET efficiency, \( E \), is calculated from this increased luminescence using
where $F_{DA}$ is donor emission after photobleaching and $F_{DB}$ is donor emission before photobleaching. Over a number of measurement sites on multiple cells, average FRET efficiencies were generally observed to be in the 10–20% range. A typical result is shown in Fig. 2(e). In this case, the average FRET efficiency across the QD-microsphere is 12%, with individual pixels ranging from 0 to 36% with a standard deviation of 10%. As anticipated, FRET was not observed on the surface areas of the microsphere that was not in intimate contact with the cell surface. Figure 2(f) shows the result of a control FRET measurement performed with the acceptor already photobleached. This demonstrates a background noise level of $\sim$4% with a standard deviation of 6.8%. Thus, the observed FRET signal is significantly above the noise level. Comparing to averaged FRET signal observed with FN-QD-microsphere (8–10% above the background noise level), the averaged FRET signal observed with BSA-QD-microsphere was significantly lower (2–5% above the background level). We interpret the FRET signal with the BSA as representing background plus free integrin on the cell surface. The approximately twofold higher signal with the FN indicates that integrin is accumulating at the bead site as a result of integrin binding to the FN.

These results demonstrate that this QD-based FRET technique can be applied to study the integrin-ECM interactions in live cells. In an in vitro study, Dennis and Bao have described FRET between QD and several fluorescent proteins, in which the QD was separated from RFP proteins by a layer of PEG lipid. They calculated the FRET distance between QD and fluorescent proteins to be 5–6 nm, with the FRET efficiency to be 26–50% for a QD:fluorescent protein ratio of 1. We measured a lower FRET efficiency, suggesting that the QD-RFP distance is larger than their system. Because the system is dynamic and the FRET pair may or may not be physically tied together, there will be a range of donor/acceptor distances sampled in this experiment. The standard relationship between FRET efficiency and an apparent or most likely distance is given as

$E = (F_{DA} - F_{DB})/F_{DA},$

where $R$ is the separation distance. For the FRET pair used in these experiments, efficiencies of $\sim$15% suggest a most likely separation of $\sim$7 nm. This is consistent with our projected QD-RFP distance of 6–10 nm, considering that cell membrane lipid-bilayer is $\sim$4 nm thick. In addition to the QD-RFP distance, the ratio of RFP:QD pairs at the local spot would also affect the FRET measurement, with higher ratio corresponding to greater FRET efficiency.

In conclusion, we have developed a QD-based FRET technique that is integrated with an AFM for detection of the cell surface integrin-ECM interactions in live cells. By employing the FN-QD-microsphere as the FRET donor, we have demonstrated the detection of integrin-FN interactions on cell surface. Because QDs are resistant to photobleaching, this novel technique also enables multiple FRET measurements. Combination of the FRET approach with the AFM has the benefits of allowing precise positioning of the QDs on the cell surface for measurement of FRET and enabling the measurement and/or application of force to the cell simultaneously with the measurement of FRET. This flexible FRET method should be applicable to the detection of other cell surface proteins and/or receptor-ligand interactions. In addition, it is relatively easy to coat the QD-microsphere with proteins through adsorption. This eliminates the need to construct a donor fluorophore-conjugated protein and thus makes the method more accessible for biological studies.

Acknowledgement
This work was supported by the National Institute of Health Grant No. R21EB005840.

References