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Distinct Migratory Properties of M1, M2, and Resident Macrophages Are Regulated by $\alpha_d\beta_2$ and $\alpha_m\beta_2$ Integrin-Mediated Adhesion

Kui Cui  
*Quillen-Dishner College of Medicine*

Christopher L. Ardell  
*Quillen-Dishner College of Medicine*

Nataly P. Podolnikova  
*Arizona State University*

Valentin P. Yakubenko  
*Quillen-Dishner College of Medicine, yakubenko@etsu.edu*

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Distinct Migratory Properties of M1, M2, and Resident Macrophages Are Regulated by $\alpha_D\beta_2$ and $\alpha_M\beta_2$ Integrin-Mediated Adhesion

Kui Cui 1, Christopher L. Ardell 1, Nataly P. Podolnikova 2 and Valentin P. Yakubenko 1*

1 Department of Biomedical Sciences, Center of Excellence for Inflammation, Infectious Disease and Immunity, Quillen College of Medicine, East Tennessee State University, Johnson City, TN, United States, 2 Center for Metabolic and Vascular Biology, School of Life Sciences, Arizona State University, Tempe, AZ, United States

Chronic inflammation is essential mechanism during the development of cardiovascular and metabolic diseases. The outcome of diseases depends on the balance between the migration/accumulation of pro-inflammatory (M1) and anti-inflammatory (M2) macrophages in damaged tissue. The mechanism of macrophage migration and subsequent accumulation is still not fully understood. Currently, the amoeboid adhesion-independent motility is considered essential for leukocyte migration in the three-dimensional environment. We challenge this hypothesis by studying the contribution of leukocyte adhesive receptors, integrins $\alpha_M\beta_2$, and $\alpha_D\beta_2$, to three-dimensional migration of M1-polarized, M2-polarized, and resident macrophages. Both integrins have a moderate expression on M2 macrophages, while $\alpha_D\beta_2$ is upregulated on M1 and $\alpha_M\beta_2$ demonstrates high expression on resident macrophages. The level of integrin expression determines its contribution to macrophage migration. Namely, intermediate expression supports macrophage migration, while a high integrin density inhibits it. Using in vitro three-dimensional migration and in vivo tracking of adoptively-transferred fluorescently-labeled macrophages during the resolution of inflammation, we found that strong adhesion of M1-activated macrophages translates to weak 3D migration, while moderate adhesion of M2-activated macrophages generates dynamic motility. Reduced migration of M1 macrophages depends on the high expression of $\alpha_D\beta_2$, since $\alpha_D$-deficiency decreased M1 macrophage adhesion and improved migration in fibrin matrix and peritoneal tissue. Similarly, the high expression of $\alpha_M\beta_2$ on resident macrophages prevents their amoeboid migration, which is markedly increased in $\alpha_M$-deficient macrophages. In contrast, $\alpha_D$- and $\alpha_M$-knockouts decrease the migration of M2 macrophages, demonstrating that moderate integrin expression supports cell motility. The results were confirmed in a diet-induced diabetes model. $\alpha_D$ deficiency prevents the retention of inflammatory macrophages in adipose tissue and improves metabolic parameters, while $\alpha_M$ deficiency does not affect macrophage accumulation. Summarizing, $\beta_2$ integrin-mediated adhesion may inhibit amoeboid and mesenchymal macrophage migration or support mesenchymal migration in tissue, and, therefore, represents an important target to control inflammation.

Keywords: integrin $\alpha_D\beta_2$(CD11d/CD18), integrin $\alpha_M\beta_2$(CD11b/CD18), macrophages (M1/M2), migration, inflammation, adhesive receptors
INTRODUCTION

Monocyte/macrophage migration to and accumulation within the site of inflammation are critical steps in the development of the inflammatory response. While acute inflammation is usually generated as a defensive mechanism, the development of chronic inflammation is an essential step in the initiation or progression of many devastating diseases including atherosclerosis, diabetes, obesity, arthritis and others (1–4). Macrophage accumulation at the damaged tissue is a hallmark of inflammation (5, 6). However, the particular subset of accumulated macrophages is critical for the further development or resolution of chronic inflammation. Classically activated (M1) macrophages produce a harsh pro-inflammatory response, while alternatively activated (M2) macrophages may have anti-inflammatory functions (7, 8). The balance between the accumulation of pro-inflammatory and anti-inflammatory macrophages regulates the fate of inflammation. So far, the mechanism of macrophage accumulation is not fully understood.

Macrophage accumulation at the site of inflammation depends upon monocyte recruitment, macrophage retention and emigration. Monocyte recruitment includes activation, diapedesis through the endothelial monolayer (2D migration) (9, 10), and migration through the extracellular matrix to the site of inflammation (3D migration). While the role of leukocyte adhesive receptors in 2D migration is well-established (9, 11), their contribution to macrophage migration through 3D extracellular matrix (ECM) is still unclear. Macrophages utilize two types of motility in a 3D environment—amoeboid and mesenchymal. Ameoboid migration is adhesion-independent movement that is based on flowing and squeezing. This migratory mode was shown to be dominant for neutrophils, dendritic cells and lymphocytes (12). Mesenchymal migration involves the classical adhesion-mediated mechanism that includes cell protrusion and adhesion of the leading edge, followed by detachment of the trailing edge and retraction of the contractile cell rear (13). It has been shown that cell-substratum adhesiveness regulates the fate of mesenchymal cell migration. Namely, an intermediate level of adhesiveness generates the optimal conditions for cell migration (14). Low adhesiveness does not support cell motility, while a very high level of adhesiveness thwarts cell locomotion because it inhibits cell detachment from the substrate (15, 16). The density of adhesive receptors on the cell surface is one of the most critical parameters of cell-substratum adhesiveness. Therefore, a high density of cell adhesion receptors that generate a high adhesiveness may lead to the retention of cells (15, 17).

Integrins are the most important cell adhesive receptors that are involved in monocyte/macrophage migration. Of particular note is the subfamily of β2 integrins that are exclusively expressed on leukocytes and consist of four members: α1β2 (CD11a/CD18), α3β2 (CD11b/CD18), α5β2 (CD11c/CD18), and αDβ2 (CD11d/CD18) (18). Integrins αMβ2 and αDβ2 are the most interesting members with regard to cell migration, since α1β2 has no ligands in ECM (19) and αXβ2 demonstrated a very low expression on macrophages (20). In contrast, αM and αD have marked macrophage expression and share many ECM ligands (21, 22).

Different subsets of macrophages have a diverse expression of integrins (23) and, most importantly, possess different migratory characteristics (24). We hypothesize that integrin expression regulates the distinct migratory properties of M1-polarized, M2-polarized, and resident macrophages. We realize that in vitro activated M1 and M2 macrophages do not fully represent the varieties of pro-inflammatory and anti-inflammatory macrophages in vivo; however, these cells are appropriate models that can help us to understand the migratory mechanisms of different macrophage subsets during inflammatory diseases.

In our previous project, we found that the pro-atherogenic role of integrin αDβ2 depends upon the upregulation of αD on pro-inflammatory M1 macrophages in vitro and on macrophages in atherosclerotic lesions, which apparently mediates macrophage retention (23). In agreement with this, αD-deficiency reduced the development of atherosclerosis and released the migration of M1 macrophages in vitro (23).

In this paper we further develop this project by analysing the role of β2 integrins on different subsets of macrophages and attempt to depict the mechanisms that stimulate cell migration/retention based on the analysis of integrin expression, cell adhesion, secretion of proteases, and mode of cell migration. We found a strong correlation between macrophage migration and expression of αMβ2 and αDβ2. A moderate expression of αMβ2 and αDβ2 on M2 macrophages supports cell movement, while the upregulation of αDβ2 on M1 macrophages and αM on resident macrophages prevents mesenchymal and/or amoeboid migration. These results were verified by using αM- and αD-deficient macrophages in 3D in vitro migration and by using an in vivo model for the resolution of peritoneal inflammation and diet-induced diabetes.

Therefore, the regulation of β2 integrin expression may help to shift the pro-/anti-inflammatory balance at the site of inflammation and reduced the pathophysiological outcome.

MATERIALS AND METHODS

Reagents and Antibodies

Reagents were purchased from Sigma-Aldrich (St. Louis, MO, United States) and Thermo Fisher Scientific (Waltham, MA, United States). Rock inhibitor (Y27632) and aprotinin were from Sigma-Aldrich. Recombinant human and mouse IFNγ, IL-4, MCP-1, and FMLP were purchased from Invitrogen Corporation (Carlsbad, CA, United States). Anti-human α1 mAb (clone 2401) was generously provided by Eli Lilly Corporation (Indianapolis, IN, United States). Polyclonal antibody against the αD I-domain was described previously (10). The antibody recognizes both human and mouse αD-I-domains and has no cross-reactivity with other integral membrane proteins.

Abbreviations: ECM, Extracellular matrix; EDTA, Ethylenediaminetetraacetic acid; FACS, Fluorescence-activated cell sorting; FMLP, N-Formylmethionine-leucyl-phenylalanine; IFNγ, interferon-γ; IL-4, interleukin 4; MCP-1, Monocyte chemoattractant protein-1; ROCK, Rho-associated protein kinase; TG, thiglycollate; WT, wild type; 2D, 2 dimensional; 3D, 3 dimensional.
with recombinant human and mouse $\alpha_M$, $\alpha_X$, and $\alpha_I$-domains. The antibody was isolated from rabbit serum by affinity chromatography using $\alpha_2$-I-domain-Sepharose. Mouse PE-cy7 and APC-conjugated anti-$\alpha_M$ mAb (clone M1/70) and F4/80 mAbs were from eBioscience (San Diego, CA, United States). The mAb 44a directed against the human $\alpha_M$ integrin subunit was purified from the conditioned media of the hybridoma cell line obtained from American Type Culture Collection (ATCC, Manassas, VA, United States) using protein A agarose (GE Healthcare, Piscataway, NJ, United States).

**Animals**

Wild type (C57BL/6J, stock # 000664) and integrin deficient mice have been backcrossed to C57BL/6 for at least ten generations. All procedures were performed according to animal protocols approved by East Tennessee State University IACUC.

**Flow Cytometry Analysis**

Flow cytometry analysis was performed to assess the expression of $\alpha_D$ and $\alpha_M$ on mouse peritoneal macrophages. Cells were harvested and pre-incubated with 4% normal goat serum for 30 min at 4°C, then $2 \times 10^6$ cells were incubated with specific antibody for 30 min at 4°C. Non-conjugated antibodies required additional incubation with Alexa 488 or PE-cy7-donkey anti-mouse IgG (at a 1:1,000 dilution) for 30 min at 4°C. Finally, the cells were washed and analyzed using a Fortessa X-20 (Becton Dickinson).

**Generation of Classically Activated (M1) and Alternatively Activated (M2) Mouse Macrophages**

Peritoneal macrophages from 8 to 12 week old mice (WT and $\alpha_D^{−/−}$, $n = 3$ mice per group) were harvested by lavage of the peritoneal cavity with 5 ml of sterile PBS 3 days after intraperitoneal (IP) injection of 4% thioglycollate (TG; 0.5 ml). The cells were washed twice with PBS and resuspended in complete RPMI media. The cell suspension was transferred into 100 mm petri dishes and incubated for 2 h at 37°C in humidified atmosphere containing 5% CO$_2$ atmosphere. Non-adherent cells were washed out with RPMI media, and the adherent macrophages were replenished with RPMI media. The macrophages were differentiated to M1 and M2 phenotypes by treatment with recombinant mouse interferon-γ (IFN-γ) (100 U/ml, Thermo Fisher) and interleukin 4 (IL-4) (2 nM, Thermo Fisher), respectively, for 4 days. Medium with IFN-γ and IL-4 were changed every 2 days or as required. The M1 phenotype macrophages from WT and $\alpha_D^{−/−}$ were labeled with red fluorescent marker PKH26 and green fluorescent marker PKH67, respectively, according to the manufacturer’s instructions (Sigma-Aldrich). The fluorescently-labeled cells were dissociated from the plates using 5 mM EDTA in PBS and used for the experiments thereafter.

**Cell Adhesion Assay**

The adhesion assay was performed as described previously (22) with modifications. Briefly, 96-well plates (Immuno 2HB, Cambridge, MA, United States) were coated with different concentrations of fibrinogen or Matrigel for 3 h at 37°C. The wells were post-coated with 0.5% polyvinyl alcohol for 1 h at 37°C. Mouse peritoneal macrophages or HEK 293 cells transfected with $\alpha_M$β2, or αDβ2 integrins were labeled with 10 μM Calcein AM (Molecular Probes, Eugene, OR) for 30 min at 37°C and washed with DMEM and resuspended in the same medium at a concentration of $1 \times 10^6$ cells/mL. Aliquots (50 μL) of the labeled cells were added to each well. For inhibition experiments, cells were mixed with antibodies and incubated for 15 min at 22°C before they were added to the coated wells. After 30 min of incubation at 37°C in a 5% CO$_2$ humidified atmosphere, the non-adherent cells were removed by washing with HBSS. The fluorescence was measured in a Synergy H1 fluorescence plate reader (BioTek, Winooski, VT, United States), and the number of adherent cells was determined from a labeled control.

**Migration of Macrophages in 3D Fibrin Gel and Matrigel**

The migration assay was performed as described previously (25). WT and $\alpha_D^{−/−}$ or WT and $\alpha_M^{−/−}$ peritoneal macrophages activated to M1 or M2 phenotype as described above were labeled with PKH26 red fluorescent dye and PKH67 green fluorescent dye, respectively. Cell migration assay was performed for 48 h at 37°C in 5% CO$_2$ in a sterile condition. An equal number of WT and $\alpha_D^{−/−}$ macrophages was evaluated by cytoospin of mixed cells before the experiment and at the starting point before migration. Labeled WT ($1.5 \times 10^5$) and $\alpha_D^{−/−}$ ($1.5 \times 10^5$) activated macrophages were plated on the membranes of transwell inserts with a pore size of 8 μm and 6.5 mm in diameter (Costar, Corning, NY) precoated with fibrinogen (Fg). Fibrin gel (100 μL/sample) was made by 0.75 mg/ml Fg containing 1% FBS and 1% P/S and activated by 0.5 U/ml thrombin. Matrigel (50%) was diluted by RPMI-1640 supplemented with 1% FBS and 1% P/S, 30 nM of MCP-1 (or 100 nM FMLP) were added on the top of the gel to initiate the migration. Migrating cells were detected by Leica Confocal microscope (Leica-TCS SP8) and the results were analyzed and reconstructed using IMARIS 8.0 software.

**Adoptive Transfer in the Model of Resolution of Peritoneal Inflammation**

Adoptive transfer was performed as described previously (23). Briefly, fluorescently-labeled WT (red PKH26 dye) and $\alpha_D^{−/−}$ (green PKH67 dye) M1- or M2-activated macrophages were mixed in a 1:1 ratio and further injected intraperitoneally into wildtype mice at 4 days after thioglycollate (TG)-induced inflammation. 3 days later, peritoneal macrophages were harvested with 5 ml PBS supplemented with 5 mM EDTA. The percentages of red and green fluorescent macrophages in the peritoneal exudate were assessed by fluorescence microscopy, multi-color flow cytometry (Fortessa X-20) and imaging flow cytometry (ImageStream Mark II, Amnis).
Adoptive Transfer in the Model of Diet-Induced Diabetes

The approach is based on previously published method (26) with some modifications. Monocytes were isolated from the bone marrow progenitors of WT and αD/− deficient mice using magnetic bead separation kit (Miltenyi Biotec, Gaithersburg, MD, United States). Monocytes were labeled with red, PKH26 (WT) or green, PKH67 (αD/−) fluorescent dyes. Red (1.5 × 10^6) and green (1.5 × 10^6) cells were mixed together and injected in tail vein of wild type C57BL6 mice fed high fat diet (45% kcal/fat) for 8 weeks. After 3 days adipose tissue was isolated, digested as described previously (26) and analyzed using FACS (Fortessa X-20, BD, United States) and imaging flow cytometry (ImageStream Mark II, Amnis).

Glucose Tolerance and Insulin Sensitivity Tests

Wild type and αD/− mice fed a high fat diet for 16 weeks were fasted overnight in a new cage containing water but no food, (~16 h). The following morning mice were weighed, and an initial blood glucose level was measured using a glucometer and from the tail vein. Glucose (2 grams/kg body weight of 20% D-glucose) was administered IP and at 15, 30, 60, and 120 min post injection blood glucose was again measured.

For insulin sensitivity test, mice fed a high fat diet were fasted for 5 h, starting at 7 a.m. (lights on). After fasting, mice were weighed, and the initial level of blood glucose measured as described above. Insulin (0.75 mU/g) was injected I.P, and the level of blood glucose was evaluated at 15, 30, 45, and 60 min.

Quantitative RT-PCR

Cellular mRNA was extracted from macrophages using the Qiagen Oligotex mRNA Midi Kit. mRNA was reverse transcribed with the iScript cDNA Synthesis Kit (Bio-Rad Laboratories, Inc., Hercules, CA, United States) and real-time quantitative PCR was performed using SYBR Green Supermix (Bio-Rad) on an MyIQ2 two color real-time PCR detection system (Bio-Rad), with the thermal cycler conditions suggested by the manufacturer. The sequences of integrin primers are shown below: αD forward, 5′-GGAGGCAATCAAGGTCAAGTA-3′, and reverse, 5′-ATCCA TTAGAGAGCTTGAAGCT-3′. αX forward, 5′-TCCGGTACG ATCAACAACAT-3′ and reverse, 5′-GGTGAAGTGAATCCGG AACT-3′. α4 forward, 5′-AAGGAACGCCAGGTTCATATT-3′, and reverse, 5′-TCACATTGTCTTTTGCTGTTG-3′. α5 forward, 5′-CAAGGTGACAGGACTCAGCA-3′, and reverse, 5′-GCTT CTTGACCACTCAAGTA-3′. GAPDH or 5S rRNA were used as an internal control (Ambion/Life Technologies, Grand Island, NY, United States).

THE LEVELS OF INTEGRIN EXPRESSION DETERMINE THE EFFECTS ON MACROPHAGE MIGRATION

Recently, we demonstrated that integrin αD is upregulated on M1-polarized macrophages but does not change on M2-polarized macrophages (23). We evaluated the potential changes in the expression of other fibrin-binding macrophage adhesive receptors during M1 and M2 polarization (Figure 2A). The
RT-PCR results demonstrated that αD is the only adhesive receptor that upregulates during M1 macrophage activation to compare with M2 subset (Figure 2B). We also detected the increased expression of integrin αX on M2 macrophages; however, the total expression of αX on macrophages is very low (20), which quashes its potential effect on macrophage migration. Therefore, the upregulation of integrin αD is the most significant modification that may affect the migratory properties of M1 and M2 macrophages.

Based on these data, further analysis was focused on integrin αD and related integrin αM, that possess similar ligand binding properties, but distinct surface expressions. The contributions of integrin α5 and αM to M1 and M2 migration were evaluated using αD- and αM-deficient macrophages. αD deficiency reduced the adhesion of M1 macrophages to fibrinogen (Figure 3A), but significantly increased cell migration (Figures 3C, left panel; 3E). In contrast, integrin αM deficiency has very limited effect on adhesion, due to its moderate expression on M1 macrophages (23) (Figure 2B), and did not demonstrate a significant effect on cell locomotion (Figures 3C,E). Both integrins, αD and αM, have moderate expression on M2 macrophages (23) (Figure 2B). The adhesion of M2 macrophages depends on both integrins, which is demonstrated in the presence of antibodies and integrin-deficient cells (Figure 3B). In parallel assays, the reduced migration of αM- and αD-deficient macrophages verified that both integrins help to support the mesenchymal migration of M2 macrophages (Figures 3D,F).

The deficiency of αD or αM may also modify the expression of other fibrin-binding integrins that can affect cell migration. To test this possibility, we evaluated the expression of α4, α5, αX, and αM on αD−/−, as well as αD on αM−/− macrophages activated to M1 and M2 phenotypes using RT-PCR. We did not detect any marked changes, except for the reduced expression of α5 and αX on αD−/− deficient M1 macrophages (Supplementary Figure 2). Clearly, these changes cannot significantly modify migration.

**αD-MEDIATED ADHESION IS CRITICAL FOR THE RETENTION OF M1 MACROPHAGES**

Inflamed extracellular matrix contains different β2 ligands, including fibronectin, vitronectin, thrombospondin, fibrinogen and others. Moreover, we recently showed that oxidative stress during inflammation may form ECM protein modifications with carboxyethylpyrole, which is also a ligand for β2 integrins (25). To verify the role of αD-mediated adhesion on cell migration, we performed macrophage migration in Matrigel, the model of
basement membrane matrix, which consists of laminin, collagen IV and proteoglycans. Notably, these proteins are not ligands for integrin αDβ2 or αMβ2. To confirm this, we tested the adhesion of αDβ2- and αMβ2-transfected HEK293 cells to a plate coated with Matrigel (Figure 4A). Both cell lines demonstrated strong adhesion to Matrigel, but this adhesion was independent of αD and αM, since anti-αD and anti-αM antibodies did not inhibit this binding. In contrast, the adhesion of αMβ2 and αDβ2-transfected cells to fibrinogen was significantly inhibited by these antibodies (21, 28) (Figure 4B). Apparently, the adhesion to Matrigel is mediated by integrins α1β1 and α2β1, which are receptors for laminin and collagen, and are expressed endogenously on HEK293 cells (29–31). To verify this hypothesis, we evaluated the adhesion of MOCK-transfected HEK293 cells to Matrigel and fibrinogen. These cells did not support the adhesion to fibrinogen, but demonstrated the same level of adhesion to Matrigel as αDβ2 and αMβ2 transfected cells (Figures 4A,B). Therefore, cells do not use αDβ2 for the adhesion to Matrigel. Accordingly, we detected a similar level of wild type and αD-deficient M1 macrophage migration through Matrigel, which is distinct to our data in αD-dependent fibrin matrix. Therefore, this result is in agreement with our hypothesis regarding the critical role of αD-mediated adhesion for macrophage retention during 3D migration (Figure 4C).

However, one of the mechanisms that affects mesenchymal migration is the secretion of MMPs that degrade Matrigel. To test the potential effect of αM or αD deficiency on MMPs secretion, M1 and M2 macrophages were incubated in 48-well plates for 24 h and the media was tested using gelatin zymography as we described previously (32) (Figure 4D). First, we found a much stronger secretion of MMPs (specifically MMP-9) in M2 macrophages in comparison to M1 macrophages. Second, we did not detect any significant effect of αD- or αM-knockout on MMPs secretion, particularly in regard to M1-polarized macrophages.

Interestingly, the robust secretion of collagen-specific MMP-9 by M2 macrophages can be responsible for the strong migration of these cells in Matrigel. The migration of M1 and M2 macrophages was performed in separate gels to avoid the effect of M2-released MMP-9 on the migration of M1 macrophages (Figure 5). In contrast, similar secretion of MMPs in WT and αD-deficient M1 macrophages allowed us to compare these two cell types in one sample. Therefore, the similar migration of WT and αD macrophages in Matrigel was not regulated by a different level of MMPs secretion, but by the lack of αD-mediated adhesion.

A HIGH EXPRESSION OF αM ON RESIDENT MACROPHAGES REDUCES THEIR AMOEBOID MIGRATION

To test the effect of high expression of other integrins on cell locomotion, we evaluated αM-dependent migration of resident macrophages. αM has a very high expression on peritoneal resident macrophages (Figure 6A). A comparable analysis of 3D migration in fibrin matrix between WT and αM-deficient resident peritoneal macrophages revealed a strong improvement in the migration of the αM−/− subset (Figures 6B,C right panel). Notably, αD-deficiency, which has a very low expression on resident macrophages (Figure 6A), did not affect macrophage migration (Figures 6B,C left panel). These results demonstrated that αM at high density on the cell surface can also prevent migration. It has been shown that resident macrophages apply the amoeboid migratory mode (24). Accordingly, the migration of WT and αM−/− in the presence of ROCK inhibitor, the inhibitor of amoeboid migration (33), resulted in a dramatic reduction in both the number of migrated cells and migratory distance (Figures 6B,C right panel). Therefore, macrophage adhesion-independent amoeboid migration can be reduced by integrin-mediated strong adhesion.
FIGURE 3 | The level of integrin expression determines the effect on macrophage migration. (A,B) Adhesion assay to fibrinogen of WT, αD−/− and αM−/− macrophages activated to M1 (A) and M2 (B) phenotypes. Some samples in (B) were pre-incubated with anti-αM and anti-αD blocking antibodies before the adhesion assay. Data are presented as mean ± SEM. *P < 0.05. (C,D) Migration assay of αD− and αM− deficiency M1 (C) and M2 (D) macrophages in 3D fibrin matrix. After 48 h, migrating cells were detected by a Leica Confocal microscope and the results were analyzed by IMARIS 8.0 software, scale bar = 500 µm. (E,F) Statistical analyses were performed using Student’s paired t-test (n = 4 per group). Data are presented as mean ± SEM. *P < 0.05, **P < 0.01.

FIGURE 4 | Matrigel does not support integrin αD-mediated adhesion and retention of M1 macrophages. (A,B) Adhesion of αDβ2- and αMβ2-transfected and mock-transfected HEK293 cells to Matrigel (A) and fibrinogen (B). The adhesion was performed as described above. Data are presented as mean ± SEM. *P < 0.05. (C) 3-D migration assay of WT and αD-deficient M1 macrophages in Matrigel. Migration was stimulated by 30 nM MCP-1 added to the top of the gel. After 48 h, migrating cells were detected by a Leica Confocal microscope (Leica-TCS SP8) (C, left panel). Scale bar = 500 µm. The results were analyzed by IMARIS 8.0 software. (C, right panel). (D) Evaluation of MMPs in culture media after macrophage adhesion. WT, αD−/− and αM−/− M1- and M2-activated macrophages were plated on fibrinogen. Media was collected after overnight incubation and analyzed by gelatin-zymography (D, right panel). The intensity of gelatin degradation was evaluated by Fuji software (D, left panel). Statistical analyses were performed using Student’s paired t-tests (n = 4 per group). Data are presented as mean ± SEM. *P < 0.05.
As we have done previously (using the model of resolution of peritoneal inflammation), to verify our results, we performed in vivo migration assays (Figures 1D–F). Our FACS data confirmed these results, since mostly M1 macrophages reside in the peritoneal cavity, while M2 macrophages emigrate during resolution (5.02 ± 0.31% vs. 2.57 ± 0.41%) (Figure 7C). The Amnis imaging flow cytometry verified the size and morphology of fluorescently labeled macrophages in the peritoneal cavity (Figure 7D).

According to our in vitro results and previous data (23) we demonstrated that αD-deficiency on an M1 background stimulated the emigration of macrophages from the peritoneal cavity, while αM-knockout had no effect (Figure 7E). In contrast, we detected an increased accumulation of αM-deficient M2 macrophages in the cavity, which demonstrates the supportive role of αM in the migration of M2 macrophages and remained consistent with our in vitro results. Surprisingly, we did not detect the same effect for αD/− macrophages. The difference between the migrations of WT and αD/− M2 macrophages was not significant (Figure 7E, lower panel).

WT and αM/− resident macrophages were isolated and tested using the same resolution of inflammation assay. After 72 h, we detected predominantly wild type cells in the peritoneal cavity, while αM-deficient macrophages emigrated (Figure 8A). This result was verified by flow cytometry. The number of red-fluorescent WT cells isolated from the peritoneal cavity significantly exceeded the number of green-fluorescent αM/− cells (Q4 vs. Q1), (Figure 8B). Based on this result, we suggest that αM serves for the supporting resident macrophage accumulation in the tissue, and αM-deficiency increases the efflux of resident macrophages.

To confirm this conclusion, we evaluated the number of macrophages in the non-inflamed peritoneal cavity of wild type and αM/− mice. Isolated peritoneal cells were stained with F4/80 antibodies and analyzed by flow cytometry to detect the percentage of macrophages. We found that αM-deficiency resulted in a twofold reduction in the number of resident macrophages in the cavity (Figure 8C). In contrast, αD-deficiency on resident peritoneal macrophages did not affect macrophage number. These data are in agreement with our in vitro and in vivo migration assays.

**αD DEFICIENCY REDUCES MACROPHAGE ACCUMULATION IN ADIPOSE TISSUE AND IMPROVES METABOLIC PARAMETERS**

To further confirm the contribution of αDβ2 to macrophage retention in the site of chronic inflammation, we used the...
FIGURE 6 | A high expression of αD on resident macrophages reduces their amoeboid migration. (A) The expression of integrin αD and αM on resident macrophages was detected with anti-αD and anti-αM antibodies, respectively, and tested by flow cytometry analysis. (B) Migration of peritoneal resident macrophages in 3-D fibrin matrix. Migrating resident macrophages from WT and αD−/− mice are shown in the left panel. The middle and right panels represent the migrating resident macrophages from WT and αM−/− mice with or without Rock inhibitor (Y27632). Migrating cells were detected by a Leica Confocal microscope (Leica-TCS SP8). Scale bar= 500 µm. (C) The results were analyzed by IMARIS 8.0 software. Statistical analyses were performed using Student’s paired t-tests (n = 4 per group). Data are presented as mean ± SEM. *P < 0.05.

model of diet-induced diabetes. The accumulation of pro-inflammatory (M1-like macrophages) in the inflamed adipose tissue is a hallmark of the inflammatory component of diabetes (26). It has been shown that αD is upregulated in the adipose tissue during diet-induced obesity (35), which concurs with the upregulation of αD on M1-activated macrophages in vitro and in atherosclerotic lesions (23). We also detected a strong expression of αDβ2 on adipose tissue macrophages of C57BL6 mice after 8 weeks of a high fat diet (45 kcal% fat) (Supplementary Figures 4A,B). To assess the role of αDβ2 and αMβ2 in macrophage migration during chronic inflammation, monocytes isolated from WT and αD−/− (or αM−/−) mice were labeled with red (PKH26) or green (PKH67) dyes, respectively, mixed in equal number and injected intravenously into mice on a high fat diet (Supplementary Figure 4C). The accumulation of adoptively transferred WT and integrin-deficient macrophages in the adipose tissue of these mice was evaluated after 3 days. The isolated adipose tissue was digested and analyzed by multicolor FACS. We detected a 3-fold decrease in the number of αD-deficient macrophages (in comparison to WT) in the visceral adipose tissue (Figures 9A,B). The result was verified by Imaging flow cytometry that confirmed the presence of labeled cells in the digested adipose tissue (Figure 9C). More importantly, it also demonstrates the maturation of labeled macrophages, since migrated cells expressed macrophage receptor F4/80 (Figure 9C, Lower panels), while injected monocytes lack this expression (Figure 9C, Upper panel). Interestingly, the deficiency of integrin αM, which did not significantly upregulate on M1 macrophages (23) (Figure 2B) had no effect on macrophage accumulation in adipose tissue (Figure 9A, Lower panel). Our previous data demonstrate that αD deficiency does not affect monocyte recruitment from circulation during inflammation (23). Therefore, these results are in agreement with our in vitro and in vivo experiments and with recently published data that αM deficiency does not affect the accumulation of macrophages during diet-induced obesity (36, 37).

The assessment of metabolic parameters of αD-knockout and WT mice after 16 weeks on a high fat diet confirm the physiological significance of our results by showing that a reduced number of macrophages in the adipose tissue of αD−/− mice improved glucose tolerance and insulin sensitivity (Figure 9D). On the other hand, the recently published data did not reveal a change in glucose tolerance test of αM-deficient mice in comparison to WT control after 20 weeks of high-fat diet, but detected decreased insulin sensitivity in skeletal muscle and liver (37).

Taken together, these results provide the link between integrin expression and potential pathophysiological functions.
Apparenty, the same integrin can support or inhibit 3D migration in tissue depending on the macrophage subset and the level of integrin expression on the cell surface.

DISCUSSION

The accumulation of macrophages at the site of inflammation is a complex physiological process that is critical for the development and resolution of inflammation. Macrophage apoptosis, proliferation and chemokine stimulation are important components of this mechanism, but the adhesive receptors that regulate the macrophage accumulation via cell migration and cell retention are the critical factors that generate the final outcome.

During the last decade, the role of adhesive receptors, particularly integrins, in the three-dimensional migration of immune cells in tissue has been questioned due to a new mechanism, the amoeboid mode of migration, being suggested (12, 38). However, recent data demonstrate that some immune cells, particularly macrophages, utilize adhesion-mediated mesenchymal migration in 3D matrices (13, 39). It has been shown that the migratory mode of macrophages depends on the environment and density of matrix (33). Previously, based on 2D models, it was suggested that cell migration is regulated by cell-substratum adhesiveness, which depends on substrate concentration, adhesive receptor density and affinity (15). This theory postulates that an intermediate level of adhesiveness (or intermediate expression of the adhesive receptors) is optimal for cell migration, while very low adhesiveness does not support cell locomotion and very high adhesiveness inhibits migration due to the prevention of the detachment of adhered cells. However, this theory was not evaluated during 3D migration in the tissue, which has more complex regulatory mechanisms and much stronger physiological implications. In this project, we tested integrins $\alpha_M\beta_2$ and $\alpha_D\beta_2$ as physiologically relevant models for studying the role of adhesive receptors during the migration of different subsets of macrophages. We discussed resident peritoneal macrophages and two subsets of monocyte-derived activated macrophages—classically activated (called M1), which can be generated by IFN$\gamma$/LPS or TNF$\alpha$ stimulation; and alternatively activated, which are produced by stimulation with IL-4 and/or IL-13 (called M2a) (7). For simplicity, we...
are calling the latter group M2. We realize that M1 and M2 activated macrophages are simplified models; and macrophages in the atherosclerotic lesion and adipose tissue may represent “mixed phenotypes.” However, these two subsets characterize the most variable difference in macrophage functional properties, and therefore, are an appropriate model for analyzing β2 integrin expression and functions in different macrophage subsets.

Our experimental approach is based on several observations. First, αD and αM share similar ligands (21, 22); second, these two integrins form a complex with the same β2 subunit, thus leading to similar integrin-mediated outside-in signaling during the interaction with the ligand; and third, the expressions of αD and αM are distinct on M1-polarized, M2-polarized and resident macrophages. We demonstrated that αD is upregulated on M1 macrophages, while the expression of αM is moderate (Figure 2) and (23). In contrast to these observations, the resident macrophages express a low level of αD, but have a high density of αM (Figure 6). At the same time, the expressions of both αD and αM integrins on M2 macrophages are intermediate (Figure 2).

Using these three subsets of macrophages, we found that 1) M2 macrophages possess much stronger migratory ability within 3D matrix in comparison with M1. 2) Integrins αDβ2 and αMβ2 are important receptors that regulate cell migration. 3) Similar to the 2D migration, integrins can support mesenchymal cell migration at the intermediate density and prevent mesenchymal and amoeboid cell migration at high levels of expression. 4) Even the adhesion-independent amoeboid mode can be negatively-regulated by a high expression of β2 integrins.

In this project, we show that strong adhesion via integrins is critical for cell retention that defines the different migratory properties of M1 and M2 macrophages. (Figures 3, 6). The analysis of αM, αX, αD, αS, and αA4 integrins demonstrates that the upregulation of αD on M1 macrophages is a major change in integrin expression during M1 activation. Therefore, αDβ2-mediated adhesion is crucial for the prevention of M1 macrophage migration. In a parallel line of evidence, we found that the lack of αD-dependent substrate (exemplified in Matrigel) eliminates the effect of αD on cell migration in this matrix (Figure 4). Importantly, αD-deficiency does not significantly change the expression of other macrophage integrins and the levels of MMP expression, which rules out the possibility for an indirect effect of αD knockout on M1 macrophage migration.

Taken together, these results propose that the accumulation of M1 macrophages at the site of inflammation is mediated by strong adhesion which promotes cell retention and the progression of chronic inflammation. In agreement with that, αD-deficiency prevents the accumulation of adoptively transferred fluorescently-labeled macrophage accumulation in adipose tissue during diabetes. The reduced number of macrophages is associated with reduced inflammation and improved glucose tolerance and insulin sensitivity in αD-knockout mice. These data correspond to our previous
results, that αD-deficiency reduced macrophage accumulation in atherosclerotic lesions and the development of atherosclerosis (23). Therefore, the upregulation of αD on pro-inflammatory macrophages during diabetes (35) or atherosclerosis (23) demonstrates a similar outcome, which is manifested in the macrophage retention at the site of inflammation and disease development. Interestingly, αM deficiency has pro-atherogenic effect on female and no effect on male mice (40). In agreement with this result, it has been recently shown that αM deficiency elevates glucose level and decreased insulin sensitivity after 16 weeks on a high fat diet. Taken together, these data confirm the opposite role of αDβ2 and αMβ2 on pro-inflammatory M1 macrophages.

In contrast, the stronger migratory properties of M2 macrophages indicate that these cells more easily leave the tissue toward the lymphatics. The increased phagocytic properties of M2 macrophages, coupled with their high migratory abilities, confirm the major function of anti-inflammatory macrophages—phagocytosis followed by efflux from the tissue. αD and αM support the motility of M2 macrophages, and therefore promote the emigration of M2 macrophages from the inflamed tissue. Interestingly, the role of αM in macrophage efflux during resolution was proposed previously (41).

The published data demonstrates that M2 macrophages may apply both locomotion modes, amoeboid and mesenchymal, which is supported by our observations regarding the αM and partially αD-mediated mesenchymal migration of M2 macrophages (Figure 3). In contrast, resident macrophages use preferentially amoeboid motility. Using ROCK inhibitor, we confirmed the preferential amoeboid migration of resident macrophages, but also demonstrated that amoeboid migration can be increased after the knockout of αM integrin, which has a high density on these cells (40). Therefore, these data propose an anchoring role for integrin αMβ2 for resident macrophages in tissue. This mechanism may be important for the normal homeostasis and mobilization the initial immune defense, which is mediated by resident macrophages. We showed that αM-deficiency reduced macrophage numbers in the non-inflamed peritoneal cavity (Figure 6). Therefore, the different immune pathologies associated with αM-deficiency can be at least partially related to the impaired resident macrophage number. Most importantly, since integrins can block (or reduce) amoeboid migration, it suggests the potential role of integrins in the regulation (particularly, inhibition) of 3D migration of other immune cells that use only amoeboid movement (for example neutrophils or dendritic cells).

![Figure 9](image_url)

**Figure 9** | αD deficiency reduces accumulation of monocyte-derived macrophages in adipose tissue and improves metabolic parameters during diet-induced diabetes. (A) WT and αD−/− (or αD+/−) monocytes were isolated from bone marrow, labeled with red (WT) or green (αD−/−) fluorescent dyes, respectively, mixed in an equal amount and injected into the tail vein of WT mice fed for 8 weeks with high fat diet (45% kcal/fat). After 3 days visceral adipose tissue was isolated, digested and analyzed using flow cytometry. (B) Statistical analyses were performed using Student’s paired t-tests (n = 4 per group). Data are presented as mean ± SEM. *P < 0.05. (C) Imaging flow cytometry. Upper panel represents the injected monocytes, isolated from WT and αD−/− (or αD+/−) mice, labeled with red and green fluorescent dyes, respectively. Middle (Q4) and lower (Q1) panels represent the labeled cells in digested adipose tissue. Channel 1 1- F4/80 represents macrophage staining. (D) WT mice (black circles) and αD−/−-knockout mice (white triangles) were fed with high fat diet for 16 weeks and glucose intolerance (left panel) and insulin resistance (right panel) were evaluated. N = 6 for αD−/− and n = 9 for WT per group. A statistical analysis was performed using Student’s t-test. Data are presented as mean ± SEM. *P < 0.05; **P < 0.01, compared to αD+/− group.
In summary, our study demonstrates the important contribution of $\alpha_\beta_1$ and $\alpha_\beta_2$ to the locomotion of distinct macrophage subsets and proposes a $\beta_2$-integrin dependent mechanism of macrophage retention in the tissue and efflux during the resolution of inflammation.

AUTHOR CONTRIBUTIONS

KC designed and performed the experiments, analyzed the data, and wrote the manuscript. CA performed the experiments and analyzed the data. NP analyzed the data and edited the manuscript. VY designed the research, performed the experiments, analyzed the data, and wrote the manuscript.

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SUPPLEMENTARY MATERIAL

The Supplementary Material for this article can be found online at: https://www.frontiersin.org/articles/10.3389/fimmu.2018.02650/full#supplementary-material


**Conflict of Interest Statement:** The authors declare that the research was conducted in the absence of any commercial or financial relationships that could be construed as a potential conflict of interest.